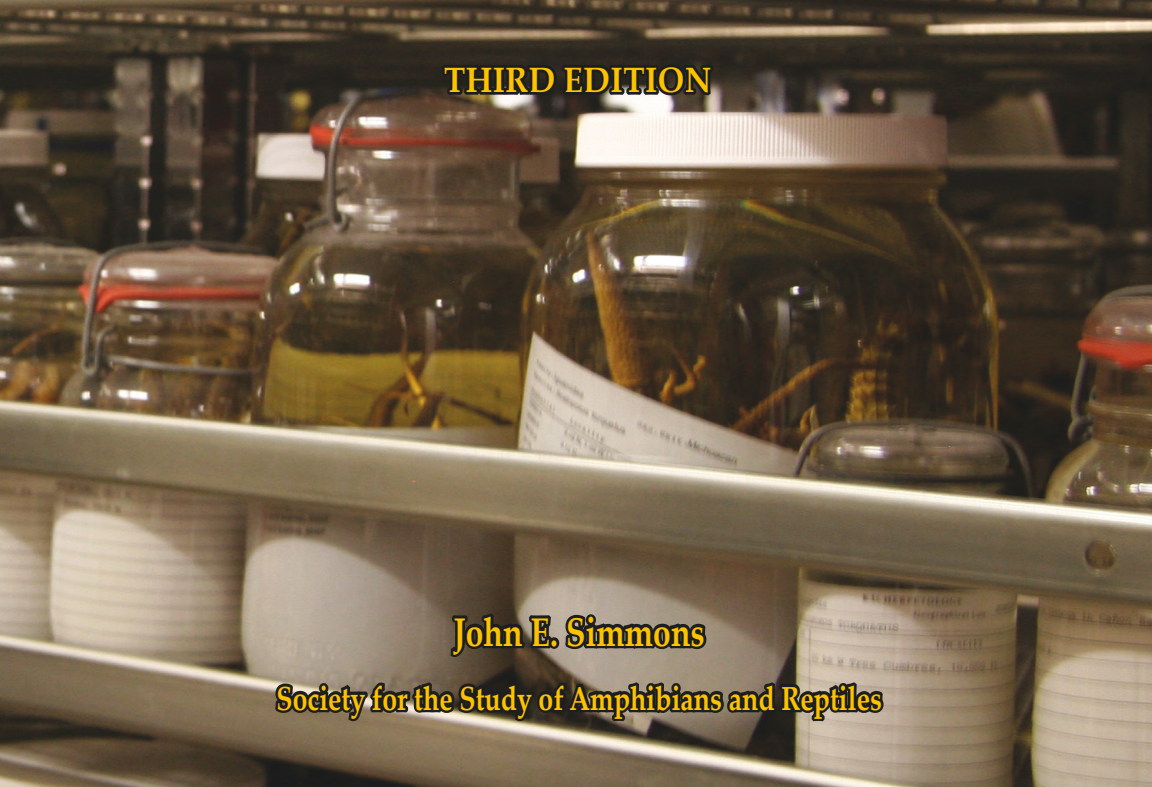




**HERPETOLOGICAL COLLECTING
AND
COLLECTIONS MANAGEMENT**

THIRD EDITION



John E. Simmons

Society for the Study of Amphibians and Reptiles

HERPETOLOGICAL COLLECTING
AND
COLLECTIONS MANAGEMENT

Third Edition

John E. Simmons

Museologica
Bellefonte, Pennsylvania
and
Earth and Mineral Sciences Museum & Art Gallery
Penn State University
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John J. Moriarty, *Editor*
3261 Victoria Street
Shoreview, MN 55126 USA
frogs@umn.edu

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Top photo - Gray Treefrog (*Hyla* sp.) adjacent to breeding habitat in Minnesota (T. Gamble)

Bottom photo - Herpetological collections of the Natural History Museum, University of Kansas, Lawrence (A. Bentley)

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INTRODUCTION

When the first edition of this publication was issued in 1987, I never imagined that nearly three decades later I would have the opportunity to produce a third edition, much less how collecting and collections management would change. The bad news is that many amphibian and reptile species have declined drastically in numbers or have gone extinct; the good news is that we now know far more about how to care for collections, which has made preserved specimens more useful for conservation, research, and teaching.

I had the good fortune to grow up when it was still possible to collect amphibians and reptiles almost anywhere, at a time when a budding young naturalist could purchase alcohol and formaldehyde over the counter at the local drug store. When I was 12 or 13 years old, I chanced upon two books that deeply impressed me—one was a novel for young people about a scientific expedition to collect animals in the wilds of China (Andrews 1951); the other contained detailed instructions on how to collect, prepare, catalog, and display natural history specimens (Brown 1954). My direction in life was set. I taught myself how to collect and preserve specimens, established my own natural history museum in a corner of the basement (Fig. 1), and began to dream of going on real collecting expeditions. Shortly after I started my freshman year at the University of Kansas, I began working part-time in the Museum of Natural History, and at the end of my sophomore year, William E. Duellman arranged for me to spend a year in the Amazon as a field assistant to Marty Crump. It was a time when populations of amphibians and reptiles still thrived, particularly in the tropics. At our field site in Ecuador, we helped record what was then the highest known diversity of amphibians and reptiles at one locality (Duellman 1978).



Figure 1. My first natural history museum.

My early memories of collecting in the tropics are of the abundance of amphibians and reptiles, of frog choruses so loud we sometimes had to shout to one another to be heard, of many nights when a few hours of collecting meant a whole day would be spent preserving and writing field notes. But it was not to last. In March of 1984, on the Páramo de Apagua in northern Ecuador, at 3480 meters in altitude, I observed a familiar sight as hundreds of little black toads, known locally as jambatos (*Atelopus ignescens*), crawled about on the damp cushion plants. Jambatos were by far the most commonly seen amphibian in the northern Andes (Fig. 2), but it was the last time I saw them in the wild. In less than five years, the species was extinct (Ron et al. 2003). It was shocking that such a common species, well-integrated into folklore and legend (Simmons 2009) could disappear so fast. Sadly, as Joseph Mendelson (2011) has so eloquently noted, it has now become impossible to duplicate the collecting success of most earlier biologists due largely to population declines and habitat loss. As a result, properly preserved, documented, and well-managed collections have increased in research value, and the need to prepare good quality specimens is greater than ever. Kevin Winkler (2014) has passionately defended the importance of continuing to collect specimens from the wild, convincingly arguing that we need more specimens, not fewer, to better understand biodiversity and the present environmental crisis, and pointing out that “Museum biological collections and the science developed from them are the foundations of our understanding of life on Earth” (Winkler 2004:455).

Not only are collections fundamental to the cataloging of biodiversity, they are necessary for understanding evolution and ecology, and are critical to developing successful conservation science measures and deciphering the reasons why so many species are disappearing. Only museum collections provide organized, documented, chronological records of biodiversity with the voucher specimens necessary to support the data. It is imperative that collections continue to be made and preserved long into the future (Gardner



Figure 2. *Atelopus ignescens* on cushion plant (Páramo de Apagua, 21 km W Pujili, Carchi, Ecuador, 3480 m, 24 May 1984).

et al. 2014). Much has been written about the many scientific uses for natural history collections (e.g., Bradley et al. 2014; Pettitt 1994, 1995; Suarez and Tsutsui 2004), but collections are also critical learning tools for producing the next generation of biologists (Cook et al. 2014).

That we now know far more about managing collections than we did in 1987 is thanks largely to the tireless efforts of the Society for the Preservation of Natural History Collections (SPNHC), the Natural Sciences Collections Association (NatSCA), and other professional organizations focused on collections care. As our understanding of the processes of deterioration and the importance of the collection storage environment has grown, so has the literature on collections care and management.

This book is intended to assist anyone interested in collecting, preserving, and working with amphibian and reptile specimens by providing a framework for establishing best curatorial practices among diverse collections. If field workers understand how collections are processed and used in museums, they will prepare better specimens; if collection users understand how animals are collected and preserved in the field, they will make better use of the specimens; and if all of us understand how collections are managed, specimens will be better utilized and preserved for the future.

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This third edition of *Herpetological Collecting and Collections Management* is dedicated to the memory of my good friend and collaborator, Tracy Hicks (1946–2014). Tracy was an artist and avid herpetoculturist who was captivated by frogs and museum collections, who was committed to interpreting science through his art, and who showed me how to see preserved specimens from an entirely different perspective. Tracy worked with great passion to preserve amphibians and reptiles in the wild, to protect the environment, to unravel the mysteries of museum collections, and to communicate his love of nature and his fascination with science to others. He was the most loyal and worthy of friends, and is greatly missed.

PART I. A BRIEF HISTORY OF SYSTEMATIC COLLECTIONS AND SPECIMEN PREPARATION

Most scientific problems are far better understood by studying their history than their logic.

—Ernst Mayr (1982)

The failure to apply scientific methodology to preservation practices is particularly intriguing in a setting where scientific methodology prevails... very few studies in natural history have applied the scientific method to evaluate preservation treatments.

—S.L. Williams (1999)

Natural history collections have played a critical (though often overlooked) role in the development of science and our understanding of the evolution of biodiversity (Winkler 2004). A study of the history of collections shows that the available means of preservation technology (the techniques and materials used to preserve specimens) has limited what specimens could be maintained in collections and what information could be obtained from them. At the same time, as the use of preserved specimens has contributed to scientific knowledge, the need for more and better specimens has stimulated advances in preservation technology (Asma 2001; Whitehead 1970, 1971). An understanding of how collecting and preparation techniques have evolved and how preservation technology has influenced research, and in turn how research needs have driven the development of preservation technology and use of scientific specimens, is fundamental to managing collections properly.

Collections have existed for much longer than museums, beginning with the basic human impulse to collect that led to the accumulation of private prestige collections in antiquity (Simmons 2010). In the Renaissance, many private collections took the form of cabinets of curiosities, dominated by objects that were valued for being rare and unusual (Lindauer 2010). During the Enlightenment, collections from these cabinets of curiosities were expanded and organized to become rational tools for understanding the world. From these ordered and carefully arrayed collections emerged the modern complex, discipline-based natural history collections that are critical for the study of biodiversity (Hooper-Greenhill 1992; Mason 2006; Murray 1904; Simmons 2010).

Natural history specimens were some of the first objects to be gathered in collections (Asma 2001; de Borhegyi and Hanson 1982; Pearce 1992), useful along with other objects to help find order in the chaos of the world and understand natural phenomena—as one author phrased it, “To collect is not to mirror the world, but to remake it” (Poliquin 2012:15), while another has asked, “Could you conceive of being human without using objects to negotiate the world?” (MacGregor 2010:19). Today there are more than 3.5 billion natural history specimens preserved worldwide in 6,500 museums (Duckworth et al. 1993; Mares 1993), far more objects than are in collections of art or history museums. One of the distinguishing features of natural history collections is that most of the specimens in them were collected to be used for research, which affects both collection growth and how collections

are managed and used. In the mid-1980s, it was estimated that worldwide, the ratio of natural history collection care workers to specimens was about 1:200,000; this ratio has become even more skewed as collections have continued to grow in both numbers and the types of specimen preparations, but few museums have added collections care positions, which means that these invaluable resources must be carefully managed at the collection level (Simmons 2013).

The history of natural history collections can be divided into eight periods (based on the six periods proposed by Whitehead 1970, 1971):

1. Ancient, up to 750 BCE
2. Greco-Roman, 750 BCE to 400 CE
3. Middle Ages, 400 to 1400
4. Renaissance, 1400 to 1600
5. Enlightenment, 1600 to 1750
6. Linnean, 1750 to 1850
7. Darwinian, 1850 to 1980
8. Post-Darwinian, 1980 to present

Ancient Collections, up to 750 BCE

The impulse to make and organize collections is a universal expression of human culture (there is evidence that Neanderthals collected objects 50,000 years ago). Ancient collections were used as visual demonstrations of wealth and prestige, but also for teaching critical skills—for example, the 20,000 clay tablets from the archive at Ebla, collected around 2250 BCE, were used to train scribes (Lewis 1992). Many ancient collections included natural history specimens, such as those of Tuthmosis III (1504–1450 BCE) in Egypt (whose collection included specimens of flora and fauna from Asia) and Nebuchadrezzar (605–562 BCE), who kept large collections in Babylon (Rigby and Rigby 1944).

Dehydration was the only means of preservation technology available in these ancient collections, so only specimens that could be dried out could be saved. A form of dehydration—mummification—is the oldest method for the intentional preservation of organic materials. Mummies were made at least 8,000 years ago by the Chinchorro culture in northern Chile and southern Peru and at least 5,000 years ago in Egypt (Arriaza 1995; Brier 1998). Ancient South Americans occasionally mummified dogs as well as humans; ancient Egyptians prepared mummies of baboons, bulls, cats, crocodiles, dogs, falcons, fish, ibis, jackals, lizards, mongooses, snakes, and other animals. Mummies were made by elaborate processes that involved removing the viscera and dehydrating the tissues. The Egyptians used natron (a naturally occurring mixture of sodium carbonate decahydrate and sodium bicarbonate) to extract moisture from tissues, then applied oils, resins, and spices, and wrapped the body in textiles. The South American mummy makers used a variety of techniques including replacing bones with wood supports, drying bodies over hot coals, coating them with ash paste and manganese, and wrapping them in pelican skins or textiles.

Collection documentation probably began not long after writing systems were developed in Mesopotamia around 3200 BCE (the oldest known writing samples are inventory lists). A specimen of a fossil sea urchin, found in

Heliopolis (Egypt), was inscribed in 1500 BCE with hieroglyphs recording the name of its collector and the location where it was collected (Murray 1904). The oldest known documented museum object is from 550 BCE, from the Sumarian city of Ur, in present day Iraq (Woolley 1952; Woolley and Mallowan 1962), where excavations unearthed a museum of antiquities that included a clay cylinder tablet that served as the object label.

Greco-Roman, 750 BCE to 400 CE

Aristotle (384–322 BCE) developed a system of classification of organisms that endured for a very long time. A good part of Aristotle's knowledge of animals came from his own dissections and observations of specimens (French 1994). Aristotle recognized approximately 540 species of animals, arranged in a graded progression or *scala natura*. The typological philosophical concept underpinning the *scala natura*—Aristotle believed in the immutability of species and that all living organisms could be arranged in order from primitive to advanced—dominated biological thought for thousands of years.

Aristotle may also be credited with starting the long tradition of sending students into the field to collect specimens from faraway places (Flower 1898), a custom that continues today. Aristotle's most famous student, Alexander the Great, collected interesting specimens he encountered while conquering the world and sent them back to his old professor. For example, Aristotle's description of elephants in *Historia animalium* reveals that he had knowledge of Indian elephants from information obtained by Alexander, and possibly had access to specimens of elephant skin and bone (French 1994). Alexander first saw elephants when he defeated Darius at the Battle of Gauqamela in 331 BCE.

The roots of the modern museum—a place where learning and objects are combined—can be recognized in the Temple of the Muses (Greek *mousetion*), founded in the 3rd century BCE in Alexandria (Egypt) by Ptolemy Sotor (305–283 BCE) (Alexander 1979; French 1994; Simpson 1998; Singer 1959). A former student of Aristotle, named Demetrius, served as an advisor to Ptolemy and helped create the museum and library, the largest in the ancient world. The Temple of the Muses continued to flourish under Ptolemy Philadelphus (285–246 BCE), its collections expanding to eventually include art objects and natural history specimens as well as manuscripts. It was a gathering place where scholars including Archimedes, Erasistratus, Euclid, and Hero taught their students while surrounded by objects, including paintings, statuary, a botanical garden, a zoo, and the library (Empereur 2002). Natural history collecting was widespread by this time—for example, an exotic sea shell collection was found in the ruins of Pompeii, which was destroyed in a volcanic eruption in 79 CE (French 1994). During this period, preservation technology continued to be limited to dehydration (Singer 1959; Whitehead 1970).

Middle Ages, 400 to 1400

The history of modern science began in China and Arabia with the development of alchemy, which reached Europe via Alexandria and other centers of learning in Hellenistic Egypt (Holmyard 1957). Alchemy had both a practical and an esoteric side. At its core was the search for a substance called the philosopher's stone that could be used to transmute base metals

into precious metals. To achieve this end, the most pure forms of the elements were believed to be necessary, which led alchemists to conduct extensive chemical experiments, yet they routinely described their work in mystical, secret illusory language that was difficult to understand. Alchemy continued to be actively practiced late into history (e.g., Isaac Newton conducted alchemical experiments).

After the collapse of the Roman Empire, most European collections were owned by churches and monasteries (Lewis 1992; Murray 1904), which were the most powerful public institutions and also the centers of intellectual activity. The collections were often enriched by exotic treasures brought back to Europe by returning crusaders and other travelers. As a result, ecclesiastical collections contained both sacred and profane objects including religious relics, paintings, statuary, and natural history specimens such as fossils, bones, and ostrich eggs. These collections were not managed in the modern sense, and were rarely used for study, but were exhibited and were significant as the forerunners of the cabinets of curiosities.

Renaissance, 1400 to 1600

Much of the work of the Greek natural philosophers was translated into Arabic and amplified by Islamic scholars as it circulated through the Near East, reaching Europe in the 12th and 13th centuries when the Arabic texts were translated into Latin (Holmyard 1990). These translations were part of the European revival of learning that produced the first cabinets of curiosities (also called *kunstkammer* or *wunderkammer*). By the mid-16th century, cabinets of curiosities could be found in many wealthy European homes and palaces, the “remnants of a rich collecting culture that flourished in early modern Europe,” the property of “rulers, patricians, humanist scholars, lawyers, physicians, and apothecaries...” (Findlen 2000:iv). What was most treasured in the cabinets were marvels—natural history specimens and other objects that were unusual or inexplicable—as these were thought to illuminate the nature of the presumed divine order in the universe.

The collections in the cabinets of curiosities were assembled at the whim of their owners, and many showed little recognizable organization into modern categories. Instead, objects were grouped in categories such as *miracula*, *mirabilia*, *artificialia*, and *naturalia*. It was from this spirit of “the veneration of the rare, the unusual, the wonderful and the miraculous” that modern museums arose (Whitehead 1970:51). Nevertheless, by the end of the Renaissance some of the functions of modern collections management began to appear, including detailed collection organization schemes and the recording of essential information about the objects (Impey and MacGregor 1985), and catalogs and object labels were being devised (Macdonald 2006), even though preservation technology was still primitive. Murray noted that “the virtue of labels” meant that “everything in the Aldrovandi museum at Bologna was—at least in 1688—described on a ticket attached to it, and the same thing was done in the Plater museum at Basle in 1663” (Murray 1904:206). Museum catalogs were initially hand-written lists of the contents of collections, but gradually became more detailed, including descriptions of individual objects and specimens. After the introduction of printing

with moveable type, catalogs took the form of printed documents that were widely distributed, some with illustrations—for example, in 1655, a catalog was published of the collection of Olaus Worm (1588–1654) of Copenhagen that was read throughout Europe (Murray 1904; Schepelern 1985).

Because many of the objects in the cabinets of curiosities were believed to have healing powers, there was an overlap between early scientific and apothecarial collections. Goode (1889) went as far as to suggest that apothecaries were the antecedents of modern curators, quoting the description of the apothecary shop found in Shakespeare:

..... Meager were his looks.
Sharp misery had worn him to the bones;
And in his needy shop a tortoise hung.
An alligator stuffed, and other skins
Of ill-shaped fishes...
(*Romeo and Juliet*. Act V, Scene 1)

Even then adequate funding to maintain the facility (“needy shop”) was a problem.

During the Renaissance, the invention of printing with moveable type and the availability of good quality paper (Basbanes 2014) played an important role in the advancement of scientific knowledge (e.g., between 1469 and 1499, at least 39 editions of Pliny’s *Natural History* and 11 editions of Aristotle’s works were published). As cabinets of curiosities continued to blossom across Europe, observation and experimentation became the established methods of scientific study (Singer 1941). Most natural history collections were arranged according to Aristotle’s *scala natura*, but as collections continued to grow, new and hitherto unknown plants and animals arrived in Europe from the Americas, Australia, Africa, and Asia that had to be fit into the classification schemes (Simmons and Snider 2012). It was in this atmosphere that the first great natural history collectors appeared, including Conrad Gessner (1516–1565) and Ulisse Aldrovandi (1522–1605). Gessner (also spelled Gesner) was born in Zurich and spent most of his working life there, assembling a natural history collection and publishing on it. Gessner’s work marked a change from the Aristotelian view of amphibians and reptiles to a modern view, particularly in his books *Quadrupedibus Oviparis* (1554) and *Serpentium Natura* (1587) (Adler 2014; Nordenskiöld 1949). Gessner’s *Historia Animalium*, a work of 3,500 pages in four immense folio volumes, included many previously unknown animals such as the opossum, bird of paradise, sloth, guinea pig, and armadillo (Ashworth 1991; Impey and MacGregor 1985). Aldrovandi, born in Bologna, was a professor of medicine in Padua and Rome. He spent his fortune collecting animals and hiring artists to draw them, and in the process founded what is widely considered to be the earliest modern natural history museum (Adler 2007). It took Aldrovandi fifty years to get the first volume of his masterwork ready for publication, but it stood as the most comprehensive zoology text until the 18th century. Bauer et al. (2013) documented 19 remaining specimens of amphibians and reptiles from the Aldrovandi collection as the oldest herpetological specimens in the world (all are preserved as dry mounts or skeletons).

Enlightenment, 1600 to 1750

During this period, museum collections proliferated and expanded, becoming indispensable for preserving the artifacts of history and the taxonomy of the natural world (Orosz 1990). The mid-17th century saw the first successful improvements in preservation technology, including the rise of taxidermy, the application of arsenic and mercuric chloride as pesticides, the use of colored wax and mercury anatomical injections, and the discovery that specimens could be preserved in alcohol (Simmons 2014; Whitehead 1971). A paper published in 1748 by René-Antoine Réaumur (1688–1757) listed the four principal techniques for specimen preparation: (1) filling or stuffing the skin and allowing it to dry (no preservatives); (2) embalming the specimen with spices, salt, alum, or lime; (3) oven drying the whole specimen; and (4) preserving the specimen in spirit of wine, or alcohol (Réaumur 1748).

The knowledge of how to produce alcohol by fermentation is very old (Forbes 1948; Simmons 2014)—there is a 6,000 year old recipe for beer from Iran (Michel et al. 1992), and the oldest evidence for alcoholic beverage production dates to at least 9,000 years ago in China (McGovern 2009). However, fermentation can only produce alcohol of about 12%, because the alcohol kills the yeast—distillation is required to make stronger alcohol. Distillation was developed in the Middle East or North Africa, but its exact origins are obscure (Holmyard 1990). Double-rimmed extraction pots that could have been used to distill alcohol were in use as early as 3500 to 3000 BCE in Mesopotamia (Brock 1993; Levey 1960). Distillation was introduced to the western world by the writings of the alchemist Zosimus in the 3rd century CE (Holmyard 1957). Distillation is based on the differences in boiling points of different liquids, and was used by alchemists to try and refine the essence, or spirit, of a substance, which is why alcohol is also called spirit of wine or just spirits (Forbes 1948). The word alcohol comes from the Arabic *al kohl*, which was a fine, black powder used as eye shadow—the fine powder was considered to be an essence, or spirit of an element (Holmyard 1957; Leicester 1956).

Another advance in preservation technology came about in 1615 when the English Crown issued a decree that gave the Royal Navy priority use of wood for shipbuilding, forcing glass furnaces to switch to coal. Coal made hotter fires, so lead oxide could be added to the glass to make an inexpensive, clear “flint glass” with fewer bubbles (Frank 1982; Singer 1959). Things placed inside these vessels could be seen without having to break the seal and open the container.

Preservation of natural history specimens in alcohol can be traced with accuracy to 04 June 1662, when William Croone (1633–1684) showed the Royal Society of London two puppies preserved in spirit of wine in a hermetically sealed glass vessel (Birch 1968; Cole 1944; Simmons 2014). The preservative was about 67% ethyl alcohol, distilled from fermented barley mash. A snake was among the earliest alcohol preserved specimens, prepared by Robert Boyle (1627–1691) who donated it to the collection of the Royal Society in 1664. Based on his own experiments, Boyle recommended changing the alcohol after initial preservation, re-distilling used alcohol, and testing alcohol strength by saturating a strip of paper in it and then trying to light it (Simmons 2014). Alcohol was by no means the only fluid used to preserve specimens. The early

instructions for preserving specimens in fluid contain recipes for a wide variety of liquid preservatives based on acids, aluminum chloride, ammonia, arsenic, burnt alum, mercuric chloride, oil of turpentine, salt, sugar, tobacco, or vinegar (Simmons 2014). Although the technique of injecting fluids into specimens pre-dates alcohol preservation, few, if any, specimens were prepared in the field by injection before the mid-1800s; rather, specimens were simply dropped into preservative in a wooden cask or barrel (glass containers were too expensive and too fragile for travel). Most of the instructions caution the collector to make slits in the specimens for the entrance of the preservative, and to change the fluid after a day or so because it becomes diluted as it extracts water from the tissues (Simmons 2014).

The first containers used to house fluid-preserved specimens were straight-sided glass cylinders made by blowing molten glass into a wooden mold. The mouth of the cylinder was covered with a piece of wood, cork, lead, or glass, and a pig or sheep bladder was stretched over it. The bladder was then coated with wax, bitumen, or a similar substance. If done right, it made a good seal, except that it had to be destroyed to remove the specimen. A portrait painted around 1730 shows Albertus Seba (1665–1736) holding a jar containing a snake preserved in alcohol and sealed with a distinctive red wax—the red wax was later used to identify Seba specimens in the collection of the British Museum (Thomas 1892).

Most museums and private collections at this time acquired specimens by purchase from sailors and travelers returning to Europe from exotic locales. Seba built up the largest natural history collection of his time largely from purchases made on the docks in Amsterdam or from commercial dealers (Adler 2014). John Tradescant the elder (ca. 1570–1638), whose collections eventually became the Ashmolean Museum at Oxford University, was able to use his royal connections to get the British navy to help collect for him. One enterprising specimen dealer, James Petiver (ca. 1663–1718) of Aldersgate in London, printed up a sheet of instructions for preserving animals in “Rack, Rum or Brandy” (rack is a shortened form of arrack, a beverage distilled from the juice of the coconut palm or a mash of rice and molasses) and urged travelers to collect for him (Stearns 1953). Nevertheless, there were some workers in this period who collected their own specimens. Maria Sibylla Merian (1647–1717) collected frogs and anuran larvae in Friesland in 1686 to observe metamorphosis, which she illustrated in carefully detailed paintings (Adler 2012). In 1699, at the age of 52, Merian moved to Surinam with her daughter for 21 months to paint insects, flowers, amphibians, and reptiles first hand. When she returned to Amsterdam in 1701, Merian sold specimens she had collected while in Surinam, including many preserved in brandy (Owens 2007). The specimens of *Pipa pipa* depicted in Seba’s *Thesaurus* (now in the Museum Adolphi Friderici collection in the Swedish Museum of Natural History) were probably purchased from Merian (Simmons and Snider 2012). Merian’s book, *Metamorphosis Insectorum Surinamensium*, was the first publication of color images of neotropical animals (Adler 2012), and also the first published depiction of *Pipa pipa* (plate 59 of that volume).

Collection care at this time was still not terribly good. As one historian noted, “William Swainson compared the storerooms [of the British Museum] with

the catacombs at Palermo, each of which was apparently opened once a year to determine how much decay had occurred and to deposit fresh material” (Barber 1980:162).

Linnean, 1750 to 1850

During this period modern systematic museums appeared in Europe and the Americas, corresponding to the increase in scientific exploration and collecting worldwide (Singer 1941). By the late 1700s, Paris had become the world center of herpetological research (Duellman and Trueb 1986). Modern zoological nomenclature began with the publication of the 10th edition of *Systema Naturae* by Carl Linnaeus (1707–1778), in 1758. As a student, Linnaeus made botanical and ethnographic collections in the far north of Sweden; later, as a professor of botany at Upsala University, he sent his students on frequent collecting expeditions. It wasn’t easy to be a student of Linnaeus—as many as a third of his students died on collecting expeditions (Nordenskiöld 1949).

The Linnean system of nomenclature became the organizing principle for natural history collections. Because Linnaeus thought that all species had been created at one time, collections were typological, with just a few specimens to represent each species, based on the *scala natura* concept of Aristotle. Rather than continuing to collect things such as “the Frightful large bat [and] Mermaid’s hand” (Whitehead 1971:155) or the “skeleton and stuffed skin of a woman who had eighteen husbands and, because she killed four of them, was hung” (Hull 2003:23), all of which are listed in the catalogs of the Oxford Museum and the Ashmolean Museum (respectively), the Linnean classification system brought order and logic to natural history collections. The ability to arrange collections in order stimulated the search for more species. Greater emphasis began to be placed on collecting, documenting, and preserving specimens, and specimens were cataloged, systematically arranged, and reported in the scientific literature.

Fluid preservation of specimens became more common, although it was still too expensive for widespread use. The anatomist Thomas Pole (1753–1829) wrote that “Preparations of almost every part are occasionally kept in spirits... as by this mode they undergo less change of appearance than by any other method of preservation, and consequently give the best idea of the natural or diseased appearance; but the expensiveness of the glass and spirits is a great inducement to the making of so many dry preparations” (Pole 1790:163). Although few dry specimens from this era remain in collections, many fluid-preserved specimens do (Ford and Simmons 1997; Hawks 1990). The techniques and standards for fluid preservation varied widely. In a handbook for collectors published in 1817, amphibians and reptiles are described as “disgusting, their haunts, loathsome and appalling, and the power possessed by numerous species of inflicting wounds, often of fatal effect, is terrific” (Graves 1817:139). The manual recommended that turtles and large reptiles be skinned and dried with alum, and later stuffed; smaller specimens could be preserved in spirits of wine containing burnt alum (Graves 1817). When he was just 15 years old, Spencer Fullerton Baird (1823–1887) wrote to the famous John James Audubon (1785–1851) to describe a bird he had shot but was unable to identify. Audubon wrote back with a request for specimens,

instructing young Baird to “Please to collect all the Shrews, Mice, (field or wood), rats, bats, Squirrels, etc., and put them in a jar in common Rum, not whiskey, brandy or alcohol. All of the latter spirits are sure to injure the subjects” (Herrick 1917:221).

Collecting and preparing good specimens of amphibians and reptiles in fluid was a complicated, expensive, and difficult operation. Alexander von Humboldt (1769–1859) lamented that on his 1799–1804 expedition to the Americas, “Sad experience taught us but too late, that from the sultry humidity of the climate, and the frequent falls of the beasts of burden, we could preserve neither the skins of animals hastily prepared, nor the fishes and reptiles placed in phials filled with alcohol” (von Humboldt 1889). Most collectors sold their specimens, either to bring in a little extra money, or as a full-time enterprise to finance travel and study. Alfred Russel Wallace (1823–1913) and Henry Walter Bates (1825–1892) financed their Amazon journey (1848–1852) by collecting and selling specimens through an agent in London, and Wallace later supported his prolonged travels in Malaysia (1854–1862) the same way.

Early collection catalogs were bound registers. The problem with a bound catalog is that it limits what can be done with the stored information, so in order to accommodate his need to continually re-arrange information as he developed his ideas about classification, Linnaeus is credited with creating a system that was widely adopted in museums—information written on sheets that could be arranged and re-arranged without re-copying—in other words, a card file. Many other innovations in collection management appeared during this period. Between about 1820 and 1840 it became common in bird collections to prepare specimens as study skins rather than as taxidermy mounts, and to tag specimens individually (Allen 1994). Fluid preservation became more common after 1845 when England repealed a steep tax on glassware that had been instituted in 1812 to support the war against Napoleon (Allen 1994).

Darwinian, 1850 to 1980

In 1859, the publication of *On the Origin of Species* by Charles Darwin (1809–1882) initiated a fundamental revolution in biology that had a tremendous effect on how specimens were collected, preserved, stored, and exhibited (Conn 1998; Hooper-Greenhill 1992; Whitehead 1971). After about 1900 museums stopped displaying all of their collections to the public, as synoptic displays gave way to naturalistic exhibits that showed specimens in an environmental context. Collecting became increasingly systematic and collections grew larger as the need to include specimens reflective of the variation within species was recognized. As science historian Robert E. Kohler pointed out in his study of natural history collecting practices in the United States, “A scientific rationale for active collecting also became compelling, as taxonomic practices became more exacting and dependent on large series collections” (Kohler 2006:111). In the late 1880s and into the 1890s, many museums sponsored their own expeditions rather than primarily purchasing collections from commercial collectors as they had done in the past. Kohler also noted that “The history of collecting in the last two hundred years is one of continual intensification: from the geographically extensive

and serendipitous collecting of the age of exploration and empire; to the extensive and intensive methods of the age of survey; and to local and highly intensive collecting in the age of ecology” (Kohler 2006:270).

In 1852, the adult Spencer Fullerton Baird, now employed by the Smithsonian Institution, published *Directions for Collecting, Preserving, and Transporting Specimens of Natural History*. Baird recommended searching for amphibians “very early in the morning or late evenings” (Baird 1852:6) and reptiles by day or “on warm moonlight nights” when “a fire or lantern may then be used to advantage” (Baird 1852:7). Baird’s instructions stated that it was preferable to preserve amphibians and reptiles in ethyl alcohol, but that large specimens could be skinned (he provided detailed instructions for skinning) and the skins treated with arsenic and dried if alcohol was not available. In addition to ethyl alcohol, Baird included recipes for Goadby’s solution (alum and salts) and a strong brine solution as alternatives. Baird wrote that “The collector should have a small keg, jar, tin box, or other suitable vessel, partially filled with liquor, into which specimens may be thrown as collected. They should be alive, or as near it as possible when this is done, as besides the speedy and little painful death, the animal will be more apt to keep sound” (Baird 1852:13). Baird also recommended opening the specimen’s mouth, making an incision in the abdomen, and injecting the preserving fluid through the anus with a syringe to ensure proper preservation.

It was during this period that fluid preservation technology became standardized, due in large part to the availability of more reliable sources of alcohol and better, less expensive glassware. The Kilner Jar (which featured a glass lid secured using a metal ring) was invented in England in the 1840s. On 30 November 1858, a Philadelphia tinsmith named John Landis Mason patented a jar with a screw-on zinc lid and a glass liner that could be screwed down tight against the shoulder of the jar. After 1858, a variety of metal clips, rings, and rubber gaskets for sealing jars became available—all of these are commonly referred to today as Mason jars. In 1882, Henry William Putnam patented a jar with a glass lid and rubber gasket held in place by a metal clamp or bail that was called a “lightning jar” because it could be sealed quickly and easily. Glass jars became more affordable after the introduction of mass production by machines that used compressed air to force molten glass into molds. Screw-on Bakelite lids became available after 1910. Another important advancement in preservation technology was an easy means to measure preservative strength by using a type of hydrometer developed specifically for alcohol by Johann Georg Tralles (1763–1822). Nevertheless, as late as the 1880s some collectors and curators were still using brine solutions to preserve specimens (due to the high cost of alcohol), and it was common to use whatever local beverage alcohol was available (Simmons 2014). William Ferguson (1820–1887), who collected in Ceylon (now Sri Lanka) between 1830 and 1887, wrote that “I now find that nearly all the frogs, especially the small ones, put into strong spirits both by Dr. Thwaites and myself when caught, have so shriveled up and got discoloured that they are nearly worthless for the purpose of identification. I find that common Arrack is the best spirit to put all kinds of frogs into when caught...” (Ferguson 1877:5). Ceylonese arrack was distilled from fermented sap of coconut flowers.

A new, more comprehensive Smithsonian publication on collecting amphibians and reptiles was published by Leonhard Hess Stejneger (1851–1943) in 1891. Stejneger's list of collecting equipment included a shotgun or pistol, dip net, fishhooks and tackle, leather gloves, cloth bags, fishing basket or tin botanical collecting box, collecting can with strap, copper tanks filled with alcohol, placental forceps (which in the field "may be carried conveniently by the side like a sword. I found one of the buttonholes of my suspenders quite the thing for this purpose"), bottle forceps, syringes and needles, stringed labels, cheese cloth to pack specimens, knife or scalpel, scissors, thread, notebook, color reference guide, and adhesive shipping labels.

Many developments took place during this period that made collecting of amphibians and reptiles far more efficient and productive, particularly improved methods of transportation—the first passenger steamboat service began in 1807, and the first steam railroad train in 1814. The late 1800s saw the development of automobiles with internal combustion engines, followed by the development of airplanes in the early 1900s. In his biography of John Van Denburgh (1872–1924) of the California Academy of Sciences, Jennings traces improvements in transportation as Van Denburgh used first railroad access and later automobiles in his fieldwork (Jennings 1997). The practice of road cruising for reptiles and amphibians on roadways at night was popularized by Laurence M. Klauber (1883–1968) (Klauber 1935, 1939).

Several other developments in this period were also critical in herpetological collecting. Plastic bags began to be used in the field the 1960s (pers. comm. W.E. Duellman). Night collecting of amphibians and reptiles became possible in the early 20th century due to the invention of easily portable, focusable light sources. Myers (2000) has documented the lack of 18th or 19th century records of the use of lights at night to collect, and has traced the development of portable light sources. According to Myers (2000), game hunters were known to be using focused beams of light by the 1890s, but in 1891, Stejneger recommended only the use of fires or lanterns for finding reptiles at night. Carbide mining lamps, which produce a bright, even light, were patented in 1900, with design improvements rapidly following (carbide lamps work by dripping water on calcium carbide, CaC_2 , to produce acetylene gas, which is ignited to produce the light). The addition of reflectors to carbide lamps produced a focused, steady beam of light. Myers (2000) reported that although dry cell batteries with incandescent lights had been available since 1898, they were too heavy and unreliable for use in the field. However, the 1900 edition of the Sears, Roebuck and Company catalog number 124 offered a variety of small, battery-powered, hand-held flashlights for sale, promising "Instant light whenever and wherever you want it" with "no smoke—no dirt—no oil—no danger." Myers (2000) concluded that it was probably not until the 1920s that G.K. Noble (1894–1940) of the American Museum of Natural History began using improved battery-operated flashlights for collecting, and speculated that "Noble... may have been the first herpetologist to effectively use lights for night collecting as a routine procedure in the New World tropics during the 1916 Harvard–Peruvian Expedition," but even so, "The concept of night collecting as an essential technique seems to have come slowly" (Myers 2000:123). Duellman (pers. comm.) pointed out that Edward H. Taylor (1889–1978) reported using a Coleman lantern (purchased in Manila) in 1912 to find

reptiles and amphibians at night in the Philippines (Taylor 1975). In any case, by 1922 the collecting leaflets produced by the American Museum of Natural History recommended carbide or electric lights as standard field equipment. A 1927 manual for collecting by Joseph R. Slevin (1881–1957) of the California Academy of Sciences lists an “electrical spotlight” as recommended equipment, and later states that “Night collecting with a hard light is very effective. An electric spot-light or carbide search-light will be found to be about the best light to use” (Slevin 1927:235). In a 1944 manual published by the Smithsonian Institution, a flashlight or “gasoline lantern that throws light in all directions” was recommended (Anon 1944:45).

Formaldehyde (HCHO) was discovered by Aleksandr Butlerov (1828–1886) in 1858 when he detected its odor while attempting the synthesis of methylene glycol, but formaldehyde did not go into commercial production in Europe until after 1889 (Simmons 2014). A German company asked a physician named Ferdinand Blum (1865–1959) to test formaldehyde’s antiseptic properties; in 1893, Blum accidentally hardened the epidermis of his own fingers, thus discovering that formaldehyde was a good fixative. Although formaldehyde was not yet in commercial production in the United States, in 1895 Leonard Stejneger mentioned using formaldehyde in Cuba, and the following year demonstrated its use for preserving plants, insects, fishes, and reptiles at a meeting of the Biological Society of Washington, noting that formaldehyde was inexpensive, compact to carry in the field, and had “...the property of preserving specimens which could not be kept in alcohol, or could not be kept in such good condition” (Lucas 1896:604). In 1911, Stejneger added an appendix on the use of formaldehyde to his 1891 paper on collecting and preserving. It is important to note that it was only after formaldehyde went into commercial production (in Europe in 1889, in the United States in 1904; see Simmons 2014) that fluid preservation became a two-step process of formaldehyde fixation and alcohol preservation, and it took several decades for the two-step process to become widely used. As late as the 1920s some collectors were still either recommending alcohol as better than formaldehyde (e.g., Van Denburgh 1922) or not recommending formaldehyde at all (e.g., Slevin 1927).

In 1881, Stephen C. Brown, an Assistant Keeper of Reptiles at the Smithsonian, became the first person with the title *registrar* in a US museum, responsible for loans, the collection storage rooms, and record keeping (Buck and Gilmore 2010). Curation standards were much simpler then than they are today—in his 1882 annual report, curator Henry C. Yarrow wrote simply that “The collection has been put in excellent order, in fresh alcohol, and provided throughout with tags of block-tin.” Until the mid-1970s, natural history collections continued to be overseen by curators and assistants, sometimes called preparators, and their duties were rather different than they are today. A 1958 memo detailing the “Schedule of Curatorial Duties” written by E. Raymond Hall (1902–1986), then Director of the Museum of Natural History at the University of Kansas, stated that the curators were to catalog and label the specimens, install specimens in the collections, check on fluid levels in containers, make sure adequate collections care supplies were on hand, check on overdue loans, and file field notes in order. However, as collections grew larger and more

complex, as the rate of collection use increased, and as more attention was paid to preserving the collections for future use, a new kind of collections care worker was needed—specialists who were better trained and more knowledgeable than preparators (Simmons 1993, 2015).

Post-Darwinian, 1980 to present

The first person to have the title of *collections manager* in a natural history museum, in the mid-1970s, was probably either Karsten Hartel (Museum of Comparative Zoology) or Susan Jewett (National Museum of Natural History). Collections managers took on the duties of the preparators, as well as the collections-care duties of the curators, and much more. In 1985 the Society for the Preservation of Natural History Collections began promoting preventive conservation-based collections management to prolong the useful life of specimens and data. Since the mid-1970s, the role of collections managers in natural history museums has become increasingly complex as the number of specimens and the variety of preparation types to be managed has grown (e.g., frozen tissue samples used in molecular systematics began to become common in herpetological collections in the 1980s).

The minimum requirements for locality data are far more specific than they once were. Beginning in the late 1990s, GPS (global positioning system) locality data became the standard. Data regarding the relationship of the individual specimen to other specimens and the habitat where it was found are becoming as valuable as the specimen itself. A modern systematic herpetology collection contains not just preserved specimens, but histological preparations, tissue samples, field notes, audio recordings, images of specimens in life and habitats, maps, and bibliographic references. All these components of a systematic collection must be linked together and carefully managed to advance scientific research.

As the amount of data that museums had to manage increased, it was only natural to turn to computers for help, beginning in the mid-1960s when the Smithsonian launched the SELGEM program (the name was a short form of “Self-Generated Master”), which was intended to be a universal cataloging program for all museums. After some successes—and some dismal failures—SELGEM and similar programs were eventually replaced by relational databases that were based on the work of Edgar Codd (1923–2003), a British scientist who worked for IBM. Truly useful and functional databases were not available for herpetological collections until the 1980s. Although the introduction of electronic databases greatly improved the management of collection records, it also diverted significant amounts of collections manager time away from collection care.

The Future of Systematic Collections

Because collection database development has been driven primarily by those using the data and specimens, not those caring for the collections, current databases are very good at sorting and retrieving information but are not yet the tools needed to properly manage collections. At best, current databases can be used to produce labels and other documents and track loans, but little else related to collections care. The best databases available today are essentially electronic versions of Linnaeus’s card files, designed primarily to

get data into the hands of collection users, which is only part of collections management. What are needed are collection databases that also incorporate information on field preservation techniques, the storage environment, collection use, collection maintenance, and integrated pest management, along with the specimen data—only then will we be able to make real improvement in how we manage collections (Simmons 2013).

The primary purpose of natural history collections is research, but if specimens and data are not properly preserved and cared for, we will have failed in the most important part of our mission. Many museums are dispensing with paper records altogether, despite the fact that we have no way to preserve electronically recorded information for the long-term future. No current means of preserving electronic data is as reliable as using acid-free paper with carbon-based ink. Tape backups, flash memory cards (memory sticks), hard drives, and “the cloud” all depend on magnetized metallic particles embedded in polymer systems, which have extraordinarily short life spans. Museums in the future will be confronted with a continual expensive and risky conversion of electronic data as each new generation of hardware and software comes and goes (Simmons 2013).

If the history of natural history collections tells us anything, it is that the future will bring new and creative uses for collections. As recently as 30 years ago, it was thought that the DNA contained in museum specimens was useless. It is now routinely extracted for research. Similar sorts of biochemical and other research uses of specimens will be developed in the future. For many years the primary function of systematic collections was amassing large numbers of specimens for the purpose of naming large numbers of species. However, the research based on collections has expanded considerably in breadth. With proper documentation, preserved collections provide baseline data for ecological and population studies. Field notes usually contain a wealth of information on the conditions under which specimens were obtained. Parameters related to the ecology of a species that are difficult or impossible to measure in the field (e.g., food selection and prey size, reproductive condition) are studied more easily and accurately with preserved specimens. New ways of using data associated with specimens will be discovered. The field of biodiversity informatics is one example—using the specimen records from multiple natural history collections, satellite images, landform, and climate data, sophisticated predictive models can be made (Krishtalka and Humphrey 2000).

The value of systematic collections as scientific resources will continue to increase. Collection growth will slow owing to worldwide environmental changes and as increasing human populations expand into previously undeveloped areas. Areas where specimens may be collected will become fewer and fewer as natural areas are degraded by the activities of humans. Fieldwork will continue to become more complicated and more costly as travel expenses rise, documentation and permit requirements increase, and specimen preparation becomes more sophisticated. Through it all, the role of well-managed systematic collections in better understanding the natural world will certainly continue to expand.

PART II. FIELD COLLECTING

The lizard can be caught with the hand, yet it is found in
King's palaces.
—Proverbs 30:28

Standards for collecting have become more exacting to meet the demands of the evolving fields of systematics and ecology. As a result, more is expected of a collector than ever before, yet opportunities to learn field methods are increasingly scarce as the number of experienced collectors declines. This section focuses on procedures and techniques for finding, collecting, preserving, and documenting reptiles and amphibians in the field.

FIELD TRIP PLANNING

Proper planning is critical to the success of fieldwork and will insure that you are not caught unprepared by lack of equipment or by climate or geographic surprises. Planning begins with allowing sufficient time to obtain the necessary permits to do the work. The process of applying for and receiving permits may take months. Permit requirements vary greatly from place to place, and may be changed often. The applicant typically needs to provide a description of the proposed project or a formal research proposal (in the appropriate language if in a foreign country), including a detailed itinerary, identification of all personnel involved in fieldwork, species desired, and number of individuals of each species to be collected. In addition to permits, supply and equipment needs must be carefully evaluated and orders placed well in advance of the planned collecting trip, and transportation arranged.

Permits

Almost every place on the planet where amphibians and reptiles can be caught now requires the collector to obtain permission before collecting. Within the United States, federal, state, or local authorities may require permits, and in some states you may be required to have a state fishing or hunting license in addition to a collecting permit. Separate permits are usually required for collecting in protected areas, such as federal or state parks, forests, or monuments. To import specimens that have been collected outside the United States, the collector must be able to show that legal permission to collect, possess, and export the specimens from their country of origin was obtained. Refer to *Appendix I* for information on obtaining permits. Table 1 lists the main types of permissions, permits, and declarations necessary to obtain specimens and to import them into the United States. It is important to remember that a specimen is defined by the United States Fish and Wildlife Service (USFWS) and other permitting agencies as a whole animal or any part of an animal—which includes tissue samples for molecular studies.

A variety of complex laws and regulations govern the collection, possession, export, and import of both wild caught specimens and specimens previously cataloged in a museum (Duellman 1999; Malero and DeAngelis 2012; Simmons 2006; and Tompkins et al. 2010). Some species are protected by special legislation, such as the Convention on International Trade in Endangered Species (CITES) or the Endangered Species Act (ESA), and

Table 1. Types of permissions, permits, and declarations for collecting.

Type of permission, permit, or declaration	Purpose
Collecting permit	Legal permission to collect and possess specimens taken from the wild. The collecting permit may specify the number of specimens of each species allowed, where the collector is allowed to collect, or where the specimens are to be deposited. Collecting permits may be issued at the local, state, regional, or federal level.
Landowner’s permission to collect	An oral or written agreement with a private landowner to collect specimens from a particular place at a particular time.
Export permit	Permission to remove legally obtained specimens from their country of origin to another country.
Import permit	Permission to import specimens of wildlife from one country into another country
CITES permit	Required to export or import specimens of CITES listed species, or move specimens of CITES listed species from one location to another within a CITES signatory country after they are accessioned.
3-177 declaration	Declaration form that must be filed electronically or in hard copy with the United States Fish and Wildlife Service as notification that specimens were imported to or exported from the United States. This form must be filed whenever animals or animal parts (e.g., tissue samples, skins, teeth) are transported in or out of the US.
Designated port exemption	Required to bring specimens of controlled wildlife through a non-designated port of entry into the United States.

must be imported into the United States through a USFWS Designated Port unless a Designated Port Exemption Permit is obtained (see *Appendix I*). The Lacey Act requires compliance with all wildlife laws (those of the United States and other countries) by requiring any specimen imported into the United States to have been legally obtained in its country of origin.

Steps in Planning Field Work

1. Determine where you want to collect, what you want to collect, and how many individuals of each species are needed.

2. Make arrangements to deposit the specimens you collect in a museum collection. Select an institution that can provide long-term care for the collection and make the specimens available to the scientific community. If you are collecting outside of the United States, you will probably be required to deposit a portion of the specimens you collect in a museum in the country of origin of the specimens.
3. Obtain the necessary permits—these may include collecting permits, permission to conduct research, export permits, and other documents.
4. If any species you wish to collect are protected, obtain the special permission required (e.g., a CITES permit).
5. Obtain the necessary supplies and field gear.
6. Collect, preserve, and document the specimens carefully.
7. When the project is completed, file the required reports with the permitting authorities.
8. Write thank-you letters to individuals who assisted you in obtaining permits and doing fieldwork.

Bringing Specimens into the United States

1. Obtain CITES export and import permits, if applicable.
2. File a 3-177 declaration (preferably using the eDecs electronic filing system). Keep a copy of the form you file for your records (printing a copy of electronically filed documents is recommended).
3. If required, enter the United States through a USFWS Designated Port or obtain a Designated Port Exemption permit in advance.
4. On your customs declaration form, declare that you are importing wildlife.
5. If you have not filed the 3-177 electronically in advance, give the 3-177 paper form to a USFWS agent (or United States Customs agent if no USFWS agent is available). You may be asked to pay a fee for the USFWS inspection (information on possible inspection fees is available on the USFWS website).
6. If presenting a paper 3-177 form, ask the agent to stamp the form and give you a photocopy of the stamped document.
7. If necessary, file an amended 3-177 form to correct errors in identifications or specimen counts later.

Ethics and the Importance of Collecting

Considering the number of preserved amphibians and reptiles already in museum collections, why are more needed? Making new collections is important for several reasons (Winkler 2004, 2014). There are many gaps in existing collections yet to be filled. Collections need to be made to support future research, particularly research utilizing new investigative techniques. In many places, amphibian and reptile populations are on the verge of disappearing entirely, making the importance of collections from these areas critical. Preserved specimens provide an ecological and evolutionary snapshot of the environment—they document the occurrence of a species at a specific place at a particular moment in time, thus specimens collected chronologically are of great value. There are still many species that are unknown, and many that have undescribed larval or juvenile forms. Specimens must be collected to serve as voucher specimens for field research or molecular samples.

The appropriate number of specimens to collect depends on the research being done. An adequate sample of specimens from a single locality for most systematic herpetological work is generally considered to be at least 20 specimens per species, plus a range of available developmental stages. Dietary studies may require much larger samples (Kovács and Török 1997).

Specimens should be collected for a purpose, not merely to add numbers to a collection—every specimen added to a collection requires an expenditure of resources to process and maintain. Carefully prepare and document each specimen collected to maximize its usefulness for research and voucher purposes. Avoid taking samples that are needlessly large (particularly of well known, common species) but remember that samples that are too small are of little use. Many permitting agencies impose arbitrary and unscientific limitations on sample sizes. Should this happen, try to convince the permit granting authorities of the need to collect adequate samples.

In general, don't take too many or too few specimens, and minimize habitat damage while collecting (e.g., restore all disturbed natural and artificial covers, don't destroy crack and crevice habitats, minimize damage to epiphytic plants). A study of reptiles living in rock outcrops revealed that the habitat damage from the use of pry bars reduced the reptile population that the habitat could support (Goode et al. 2004).

Under normal circumstances, it is highly unlikely that scientific collecting will damage a wild population. The mortality rate for wild living amphibians and reptiles is very high, and the number removed for an adequate scientific sample should have little effect on a wild population, unless that population is already perilously low in size and restricted to a small, isolated geographic area.

Specimens should be handled carefully and euthanized in a humane manner, while still rendering them fit for good preservation. Record all data possible for each individual specimen that is collected.

Comply with all laws and regulations in effect in the area where collecting is done. Violations of the law—particularly intentional violations—will result in serious problems for the collector, for the institution in which the specimens are later deposited, and will reflect badly on the profession as a whole.

For further discussion of collecting ethics, refer to Anonymous (1987); Association of Systematics Collections (1993); Edson 1997; Herrera-MacBryde (1986); Loftin (1992); and Pearson (1986).

What to Take in the Field

Each field trip has its own supply and equipment requirements. For guidance in selecting what you will might need, see *Appendix II*.

MAKING THE COLLECTION

Bad work in collecting is, in nine cases out of ten, due to one of two causes—ignorance or laziness. By some curious process of reasoning, many really intelligent men conclude that they can go into the field and collect successfully without having learned a single thing about methods, or asked a word of advice from a competent instructor.

—W.T. Hornaday (1905)

When to Collect

When to collect is often dictated by scheduling restrictions, species desired, or habitat type. Depending on what species are desired, the best time to collect might seem to be simply an issue of day vs. night or wet season vs. dry season, but collecting at the presumed wrong times can often yield animals otherwise overlooked. Don't let preconceptions limit the choice of collecting time—take along appropriate equipment and field clothing to be prepared to work day and night, rain and shine. In general, for seasonal conditions, spring or wet-season collecting is more profitable than summer or dry-season collecting, although in many areas the dry season has the effect of concentrating wildlife around dwindling water resources, thus increasing the chances of obtaining some species. In tropical regions, some species breed primarily during the dry season. Some species shift their diel activity patterns depending on daytime and nighttime ambient temperatures.

What to Collect

Most field collectors concentrate on adult animals to the point that juveniles and particularly neonates of most amphibian and reptile species are rare in collections (Alberch 1985). Efforts should be made to collect all life stages of the species desired, including eggs and larvae, neonates and immatures, and a balance of adults of both sexes. Larvae should be raised in the field, if possible, with individuals preserved at each stage to prepare developmental series (see *Eggs*). A small effort in the field will result in very valuable collections for future research.

When preparing for a collecting trip, take the time to carefully examine museum records from the area where you are going so that you can avoid collecting animals that are not needed. In many instances, ecological monitoring and recording of the presence of species may be more valuable than making collections (e.g., Lips et al. 2001).

Sometimes the only evidence available to document the occurrence of a species in a particular area are tracks, calls, or shed skins (Fig. 3). Physical ichnospecimens (such as shed skins) should be collected and treated as other specimens; tracks can be photographed or cast in plaster; calls can be recorded.

How to Collect

In the field, once you have donned appropriate field clothing (see *Appendix II*), equip yourself with:

- a variety of plastic bags
- several cloth bags
- a notepad and pen or pencil



Figure 3. Shed skin of *Vipera berus* (Scaleby Moss, near Carlisle, England).

- a series of sequentially numbered tags to drop in the collecting bags along with the specimens to help keep your notes straight (these are not the same as your field catalog tags)
- a hat
- a thermometer for determining air and water temperature
- a snake hook, tongs, or placental forceps (with serrated ring jaws) if you intend to collect serpents
- a large knife or machete
- compass or GPS device
- for night work, a headlamp (with extra bulb and extra batteries)

When you collect an animal, drop a numbered tag in the collecting bag with the specimen. Use this number to record such data as time, temperature, habitat, and other pertinent information on your note pad—writing down data at the point of capture will ensure greater accuracy in your field notes, particularly if multiple collectors are working in the same area (Fig. 4). This information will later be incorporated into your field notes. Keep bags of specimens out of direct sunlight and away from heat; put a few damp leaves or some moss in the bags with the specimens to help them avoid dehydration.

How Much to Collect

How many specimens to collect must be a balance of the limitations imposed by permit restrictions, ethical considerations (see discussion above), adequate sample size, and specimen preparation time. If several collectors are working in an area, it is very easy for the tide of incoming specimens to overwhelm the preserving and cataloging process, which will result in the collecting of unnecessary specimens and poor specimen preparation.



Figure 4. David McLeod recording data at the point of capture (Khao Luang National Park, Thailand).

How Much Time does it take to Prepare Specimens?

As a general rule, for standard preparations, including note taking and specimen preservation, allow an average of 10 to 20 minutes per specimen. Add another 10 to 20 minutes per specimen for extraction and preparation of tissues for molecular work, and an additional 10 to 15 minutes to photograph the specimen. Preparation time speeds up when processing many specimens of the same species, but slows down when preparing fewer individuals of a variety of species. Add to this calculation approximately 30 minutes to set up the preparation lab, and another 30 minutes to clean up and break the lab down when preparation is completed. Finally, add at least 45 to 60 minutes per day for writing your field journal and notes.

Places to Collect

Never collect without permission. Ask permission of local landowners before entering private land, and make sure those in charge of public lands know that you have permits in advance and that you will be collecting. When collecting near international borders or at night near populated areas, it is a good idea to alert local law enforcement about your planned activities—walking around with a headlamp at night may otherwise seem very suspicious.

Collecting may be associated with a particular project, such as obtaining voucher specimens for an ecological study, or specimens selected to monitor reproductive activity of organisms in a community—for this type of collecting, the locality is dictated by the requirements of the study. For biodiversity surveys, when a sample of all species in an area is desired, collecting should be approached with an open mind. Sometimes the most unlikely looking places turn out to be prime habitat (Fig. 5). By day, search



Figure 5. A breeding pond for Natterjack Toads, *Epidalea calamita* (Ainsdale Sand Dunes, England).

under rocks, logs, and pieces of metal or wood lying on the ground (always restore ground covers, and minimize disruption to the environment as you work). Walk paths and trails slowly to spot animals that are actively hunting or sunning themselves. Look beneath the loose bark of dead trees, and rake through leaf litter. Use traps, drift fences, and shelters if you are in an area for a reasonable length of time. By night, walk paths and trails with a light. Search the margins of ponds, streams, and swamps (try to familiarize yourself with the area you plan to collect while it is still daylight). Many good aquatic sites can be located at night by tracking calling frogs. Cruise roads slowly in a vehicle with headlights on low beam.

Many animals that are difficult to collect during the day, such as some species of lizards and snakes, may be plucked easily from their perches at night (Hoffmeister 1951). Most collectors are familiar with the edge effect phenomenon—animals are often more plentiful at the edge of disturbed situations than they are in pristine habitat. Hunting along roads, fence rows, around human habitations, trash dumps, wells, irrigation ditches, and the like is often quite productive. In urban areas, check parks, trash piles, and even inside sunken utility meter boxes (McCoy 1961). Alert people to your activities and collecting needs. Local people, particularly children, often know exactly where to find the animals you need.

Following are some typical situations in which reptiles and amphibians may be found.

Areas being Burned or Cleared. Many species of wildlife flee from encroaching fires. Watch for movement just ahead of the heat and flames, while keeping a cautious eye on the fire itself. Following bulldozers or

tractors as land is cleared or plowed will often yield species otherwise difficult to find.

Caves. Very few amphibians and reptiles are found in caves, other than those that live near the cave mouth, although a few species of salamanders occur only in caves (Brandon 1971; Fenolio et al. 2014). However, in Thailand, I observed rat snakes (*Orthriophis taeniurus*) crawling up the walls and across the ceiling to feed on bats in Kao Surakan Cave in Nakhon Si Thammarat, deep in the dark area of the cave, far from the entrance. On the cave floor beneath the snakes and bats, toads (*Phrynoidis aspera*) were feeding on crickets that were in turn feeding on bat guano.

Cavities. Holes in trees, rocks, and stream banks as well as water collected in agaves or bromeliads (McCracken and Forstner 2008) and other plants are all good places to search for small reptiles and amphibians. Eggs or larvae are sometimes found in cavities in bamboo, hollow trees, or epiphytic plants. In El Salvador, I once saw a *Leptotyphlops* crawl out of a hole in a pinecone when it was kicked.

Deserts. By night, drive slowly along roads (see *Road Cruising*) and look around rocky areas with a light. By day, look beneath rocks and debris; search the cracks in exfoliating boulders or exposed shale, and the gaps between rocks (Morrison and Tanzer 1966). During and just after desert rains (when creatures run riot at temporary pools) may be the most productive time to collect, and may be the only time you will find most desert dwelling amphibians. Desert collecting may be improved by trapping (Banta 1957; see *Traps*). On fine-textured soils, snakes and lizards may be found by following their tracks (Bider 1968; Klauber 1944; Lillywhite 1982; Mosauer 1933). See also Whitlock (1971).

Hillsides and Pastures. Check beneath any shelters and covers, such as rocks, logs, and pieces of wood and metal. This is usually more successful on slopes of 45 degrees or less, particularly when the slope is oriented toward the sun. Thinner, flatter rocks generally are preferred over thicker ones. Be alert for eggs as well as moving creatures beneath the rocks, and always return the covers to their original position. If long-term work is planned in an area with open hillsides or pastures, artificial shelters or covers made of wood, metal, or plastic may be set out to attract amphibians and reptiles (see *Shelters and Covers*).

Leaf Litter. Sift systematically through leaf litter with a small rake, snake hook, or machete by day. Keep an eye out for amphibian and reptile eggs. In some tropical regions, diurnal leaf-litter frogs can be located by listening for calls.

Oceans. Sea snakes may occasionally be caught in nets in the open ocean, found in tide pools, or be washed up on the shore. A few species come ashore intentionally. Sea turtles are notoriously difficult to collect except when ashore, unless they are accidentally taken in fishing nets. Both Humboldt (1896) and De Sola (1932) described how sea turtles are sometimes caught by hauling them in on a line tied to a remora or sharksucker (Echeneidae).

Ponds and Lakes. Shorelines should be worked with nets (see *Nets, Seines, and Netting*) by day and searched with an artificial light source by night (see *Headlamps and Spotlights*). Be alert for animals in the water and those coming down to the water's edge. Traps for amphibian larvae and adult reptiles and amphibians are recommended (see *Traps*). Check man-made structures at ponds and lakes (Hawken 1951). Large frogs may be taken on lures or flies (Miller 1972). A plunge tray may be used to collect organisms in the water (Karns 1986).

Road Cruising. This classic technique for snake hunting in the Age of the Automobile is very effective (Klauber 1935, 1939; O'Shea 1992; Shaffer and Juterbock 1994). If seldom traveled roads are available, particularly asphalt roads, cruising at slow speeds beginning at dusk is a good way to find animals that crawl out on the road for warmth. Be prepared to leap from the vehicle quickly, and take a light with you. Exercise care when road collecting to avoid traffic, particularly in places where approaching traffic may not be clearly visible. A spotlight may be used to search for creatures on the sides of road cuts (a variety of powerful lighting devices that operate using a car battery are available, but note that their use is illegal in some jurisdictions). Road cruising is illegal in some areas, so check local regulations first. A scoop device has been described for rapidly retrieving animals off the road (Sievert et al. 1999).

Roadside Ditches and Temporary Pools. These ephemeral habitats often harbor amphibians and reptiles, particularly immediately after a rainfall. Many amphibians breed only in ephemeral pools.

Streams and Rivers. Night collect streams and rivers by walking along the bank or in the water with an artificial light source (see *Headlamps and Spotlights*). Check exposed rocks and overhanging vegetation as well as stream banks and in the water. By day, check under rocks in the streambed, use dip nets or seines, and set out aquatic traps (see *Traps*). Animals may hide in cavities created by streams that undercut their banks—for example, frogs of the genus *Telmatobius* may be collected during the day by reaching up under overhanging stream banks (Fig. 6; also see *Noodling*). Amphibian larvae blend in well with stream bottoms and hide among the gravel, leaves, and detritus. Some have sucker mouth parts that allow them to clasp the undersides of rocks, and can be caught by a downstream net when rocks are moved (Svihla 1959).

Streetlights, Porch Lights, Building Lights. Outdoor lighting attracts many insects, which in turn attract a variety of amphibians and reptiles.

Swamps. Bogs, swamps, and seepages are best collected for most species at night with an artificial light source (see *Headlamps and Spotlights*), or worked by day with nets. Swamps and bogs are excellent places for aquatic traps for amphibian larvae, turtles, and other animals (see *Traps*).

Termite Mounds and Ant Nests. Snakes, amphisbaenians, and some amphibians may be found in the soft earth at the base of termite mounds; it is easier to find them if you can first tip the termite mounds with a tractor or



Figure 6. Undercut stream bank habitat for *Telmatobius* (11.5 km SE Gualaceo, Ecuador, 2940 m).

with a lever (Pramuk and Alamillo 2003). Many tropical ant species construct large ground nests of loose material that harbor small snakes and frogs, and in parts of the United States, *Gastrophryne* may share burrows with tarantulas.

Tree Falls. Both natural tree falls and those resulting from human activity often yield seldom-collected species. Check carefully in the roots of fallen trees as well as on the trunks, under the bark, and in the upper branches.

Tree Tops. For the adventurous and the well-insured, the canopy of the forest can be accessed using modified rock-climbing techniques and lots of rope. Canopy level bromeliads and other epiphytic plants are particularly good places to look for amphibians and reptiles. A technique has been developed for using drift fences in trees (Vogt 1987). For the thrilling details of collecting in the treetops, refer to Mitchell (1982); North (1943); O'Shea (1992); Perry (1978); Tucker and Powell (1991); and Whitacre (1981).

Tropical Forests. By day, search beneath logs, under loose tree bark, and sort through leaf litter. Large tropical trees may have buttresses that fill with water and form breeding sites for amphibians. Myers (1956) described a method of catching small lizards on the forest floor by dangling an insect tied to a thin line on the end of a pole in front of likely looking spots among leaf litter and debris. Bushes may be beaten for snakes and lizards. By night, walk trails and paths using an artificial light source (see *Headlamps and Spotlights*). Finding amphibians and reptiles in tropical forest vegetation can be quite overwhelming at first—it takes time to form a search image of your intended prey. Give yourself time to adapt to the conditions. Remember to look up into

the trees and down at the forest floor as you move along trails, and not just search at comfortable eye level. Steep-sided trenches may be dug or pitfall traps placed when the collector will be in residence for a lengthy period of time (see *Pitfall Traps and Trenches*). Always cave in or fill in trenches and holes from pitfall traps when collecting in the area is completed.

Collecting Techniques and Equipment

When traveling to remote areas or out of the country to collect, never assume that you can purchase any supplies locally unless you know you will be passing through a large city and have plenty of time to shop. The simplest items can be the hardest to locate (see *Appendix II* for more information on what to take with you, and *Appendix III* for sources of supplies and equipment).

Bags. Plastic bags are recommended for amphibians and small reptiles. You can never have too many with you. Buy two mil (0.002 inch) plastic bags with long necks that can be easily tied shut. I prefer 6 x 15 inch and 8 x 18 inch bags. Avoid zipper bags (e.g., Zip-Loc) and wire clamp bags (e.g., Whirl-Pac), as it is difficult to get sufficient air in these bags for the animals to breathe, the bags are not easy to carry once the animal is inside, and the air may escape easily from them. I recommend wearing a belt while collecting so that bags may be tucked under the belt for safe keeping (leaving both hands free), or put the plastic bags in a cloth bag that is tied to the belt. Put a bit of damp leaf litter or moss inside each bag for moisture. Plastic bags may be reused several times if washed thoroughly (with water only, no soap) unless they have had a particularly noxious animal inside (e.g., certain amphibians with highly toxic skin secretions). Cloth bags should be made of white cotton or muslin that has been double washed to remove the sizing, sewn with double seams, and be deep enough to be safely tied with a venomous snake inside (see Huheey 1964). I recommend cloth bags of approximately 12 by 24 inches and 18 by 32 inches. The proper technique for bagging a venomous snake is to (1) drop the snake in the bag, (2) quickly twirl the top of the bag (to keep the snake in the bottom) and lay the bag on a flat surface, (3) confine the snake in the bottom of the bag by holding the shaft of a snake hook across the bag, and (4) tie the top of the bag tightly.

Bait. Some species of amphibians and reptiles may be lured to areas where they are easy to catch by using food bait to attract the insects that they feed on or to attract the animals themselves (Wilson and Pine 1974). Insects or fruits may be tied by a thin thread or monofilament line to a pole and dangled before an amphibian or lizard. Grab the beast when it grabs the bait (Myers 1956; Serena 1980). Small hooks baited with live insects may be dropped down the burrows of salamanders (Mount and Schwaner 1970). Bait may also be used with adhesive traps (see *Traps*).

Blowguns. These devices are recommended for diurnal collecting. Blowguns can be made from a smooth tube using protruding needlepoint darts (Hoddenbach 1966; Karns 1986; Tinkle and Lawrence 1956). A commercial blowgun (60 inches long, 40 caliber, made of Teflon-lined aluminum tubing) has also been recommended (L. Grismer, pers. comm.). The commercial blowgun fires plastic stunplugs.

Drift Fences. Drift fences (Fig. 7) are used in conjunction with shelters (see *Shelters and Covers*), pitfall traps, trenches, and funnel traps (see *Traps*). Drift fences are useful devices when a collector is in an area for an extended stay. Traps and trenches must be checked regularly, several times each day. Drift fences may be made of wire mesh, metal, cloth, plastic, or wood.

General. Campbell and Christman (1982); Dodd and Scott (1994); Enge (1997); Gibbons and Semlitsch (1982); Gibbons (1983); Karns (1986); Middendorf (1979); O'Shea (1992); Vogt and Hine (1982). Smith (1971) described the use of electric fences; Malone and Laurencio (2004) described the use of polyethylene drift fences.

Aquatic Drift Fences. Lutterschmidt and Schaefer (1996).

Arboreal Drift Fences. Vogt (1987).

Drift Fences for Amphibians. Arntzen et al. (1995); Corn (1994); Dodd (1991); Dodd and Scott (1994); Gibbons and Bennet (1974); Moulton (1954); Murphy (1993); Storm and Pimentel (1954); Sutton et al. (1999).

Drift Fences for Reptiles. Cockburn et al. (1979); Fitch (1951); Hall (1970); Milstead (1959).

Eggs. Reptile and amphibian eggs should be hatched under field conditions, if possible. Preserve reptile hatchlings immediately. Reptiles may be encouraged to deposit eggs if appropriate nesting materials are provided (Gordon and Tinkle 1960). Reptile eggs can be incubated in their native soil in a closed plastic bag or clean plastic container, opened occasionally for air; in a 1:1 mixture (by weight) of sterile potting soil (vermiculite) and distilled water (Tryon 1975); or in moist sterile cotton (Legler 1956). Amphibian eggs may be incubated in a plastic bag or clean plastic container, with water changes as necessary. An amplexant pair of amphibians may be kept in a clean plastic bag until eggs are deposited. Once the eggs hatch, raise the



Figure 7. Drift fence (Sakaerat Environmental Station, Thailand).

larvae through metamorphosis, preserving one or two at each developmental stage (for stages, refer to Duellman and Trueb 1986). Feed the developing amphibian larvae on pond detritus or tropical fish food.

Elastic Bands. Big rubber bands are extremely useful for stunning small lizards or field companions long enough to grab them. Dundee described a gun stock to fire elastic bands with greater force (Dundee 1950). See also Brown (1946) and Neill (1956). Elastic bands can also be used on frogs at night and on roaches in hotel rooms.

Electrical Devices. The use of electrical apparatus to stun or shock reptiles and amphibians in the water or in the ground have been proposed by Anderson and Smith (1950); Gunning and Lewis (1957); Harris (1965); Joanen and Perry (1971); and Smith (1971).

Forceps. Long forceps (bottle forceps) are very useful for getting animals out of cracks and holes, and for handling preserved specimens. Purchase a pair 12 inches or longer. Scissor-grip placental forceps with serrated ring jaws are also quite useful, particularly for small venomous snakes. See *Appendix III* for suppliers.

Gigging and Gaffing. A long pole with a sharp point or points may be used to collect a variety of amphibians and reptiles (Lagler 1943; Walden 1970). In some areas gigging is the only legal means to collect game species of frogs, however, it is not approved as a humane collecting technique (Anon. 1987).

GPS Device. A global positioning system (GPS) device that takes bearings from satellite transmissions is an indispensable field tool for obtaining precise locality data and for navigating your way back to camp after you get lost. Prices vary widely; desirable features include parallel channel tracking, fast signal acquisition time, memory capability, and a clear, easy to read display.

Grabbers. Several grabbing devices have been described for use with small, nimble animals. See Bedford et al. (1995); Durtsche (1996); and Witz (1996).

Headlamps and Spotlights. The use of eye-shine is indispensable when collecting at night. It is particularly useful in dense habitats, along road cuts, on cliff faces, and on walls. To spot an animal by eye-shine, use a light source with a focusing lens that can be positioned close to the level of your eyes. There are a variety of battery powered headlamps on the market; LED light sources are particularly bright. What size headlamp to use is a trade-off between weight, availability, battery life, and cost. The higher the lumen rating, the brighter the headlamp beam will be. I prefer a headlamp with at least two brightness settings, a low-level setting for walking to and from collecting areas (to save battery power) and a high-level setting for searching for animals. Mark the batteries and switch battery sets every hour to prolong battery life. See also Corbell and Fellers (2001); O'Shea (1992); and Whitaker (1967b) for advice on using eye-shine to locate animals.

Jars and Vials. Small plastic jars and vials with screw-on lids (snap-on lids come loose too easily) make handy containers for small reptiles and

amphibians, and may prevent the animal from being crushed as can happen with plastic bags. Jars and vials can be reused if washed thoroughly with water.

Local Residents. If you will be in one place for a reasonable length of time, it is often profitable to offer remuneration for specimens. Resident children are often better local collectors than the visiting field person. Herpetologists have even gone so far as to broadcast their desires for specimens over the local radio station (Shaw 1982). Keep in mind that careful questioning will be necessary to obtain the requisite information concerning the location and habitat of the individual specimen when someone else collects it. Price adjustments may have to be made over time to discourage over-collecting of common species. Other local life forms may also aid in the location of desirable specimens, as Smith (1946) discovered with flocks of snake-finding West Texas turkeys.

Machete. Very useful in thick vegetation for cutting trails, poles, and yourself, if you are not careful. Machetes can also be used for digging holes and raking through leaf litter. Purchase a machete with a sturdy scabbard that can be worn on your belt. Take along a good file to keep an edge on the machete blade.

Nets, Seines, and Strainers. Dip nets (of various sizes) and tea strainers are useful for collecting amphibian larvae (Fig. 8). Dredges and seines are also useful (Goin 1942). Meylan (1989) recommended a 6–10 meter bag seine. Bragg (1951) described a straining device for collecting aquatic organisms using a wooden frame and screen. Nets for collecting non-aquatic species have also been described for lizards (Bouskila 1985; Paterson 1998) and turtles (Lagler 1943; Recht 1981; and Vogt 1980).

Noodling. This ancient technique involves reaching under water with bare hands to grope around for an animal, then grabbing the beast (Karns 1986; Lagler 1943; Plummer 1979). It is a particularly useful technique for working undercut stream banks. Walking barefoot in the mud of ponds and swamps has also been used in the Venezuelan llanos to locate both caiman and anacondas (S. Manness, pers. comm.).

Noosing. When collecting in Sitáwaka, Ceylon (now Sri Lanka) in the mid-1800s, William Ferguson described a snake collecting implement made by a native collector that consisted of a three to four foot long bamboo pole with a running noose made of twine fixed to a notch at the end—the noose was slipped over the head of a snake and the twine pulled to tighten the noose against the bamboo (Ferguson 1877). Noosing is now a standard technique for collecting diurnal lizards, but is also used for other reptiles and occasionally for amphibians (Karns 1986). Use a slipknot of monofilament, thin thread, or dental floss and a long, thin pole. Work the noose slowly over the animal's head, then jerk upwards with a quick motion. Variations include dangling bait or a rubber band lure in front of an animal or in front of its lair to lure it to the noose (Peaker and Peaker 1967). Strong et al. (1993) described a baited noose. The noose may be stiffened by waxing the line or the noose may be made of copper wire (Eaken 1957). Nooses may also be shot from a noosing gun (Bertram and Cogger 1971), used with snap snares (Stickel 1944; Bennett



Figure 8. David Hillis using dip net to collect anuran larvae (Páramo El Angel, Carchi, Ecuador, 3150 m).

et al. 2001), or noose traps (Bennett et al. 2001; Vyas 1990; Ziegler 1999). Nooses are useful for removing animals from crevices (Morrison and Tanzer 1966), and may be used on salamanders (Camp and Lovell 1989) and skinks (Durden et al. 1995).

Poles. These are useful for several purposes, including noosing (see *Noosing*). By night, frogs and other animals perched in high bushes or trees may be coaxed onto the end of a pole and then lowered to the waiting hands of the eager collector. A long pole can be used to dislodge an animal from a high perch. A stout pole can be used as a walking stick in steep terrain, in mud and muck, and to help the collector move in swift stream current. Carpenter (1955) located turtles in piles of leaves and debris by gently probing with a thin pole. Collapsible, telescoping poles are available, and of course in wooded areas one can always be cut on the spot.

Rakes. A small rake, particularly a potato rake, is useful for combing through leaf litter, flipping objects, combing through aquatic debris, and locating aquatic turtles buried in soft sand under shallow water (Meylan 1989).

Scoops. These are used to collect animals from roadways, leaf litter, and other situations (Brattstrom 1996; Sievert et al. 1999).

Shelters and Covers. Shelters and covers are objects (such as pieces of wood, metal, or plastic pipe sections) that are positioned for animals to hide under or inside. A pitfall trap may be used beneath a shelter. Examples of how a temporary shelter may be used include tossing a hat on the ground

and allowing lizards to run under it (Klein 1951); placing cardboard tubes (with one end closed) on the ground (Strong et al. 1993); or setting out PVC pipe tubes for frogs (Bartareau 2004; Leach 2011; Moulton et al. 1996). For more information on shelters and covers, refer to Carlson and Szuch (2007); DeGraaf and Yamasaki (1992); Fellers and Drost (1994); Fitch (1992); Grant et al. (1992); Hoffman et al. (2009); Moore (2005); Parmelee and Fitch (1995); Scheffers et al. (2009); Strong et al. (1993); and Sutton et al. (1999).

Shooting. Regulations make the use of a firearm complicated in many places, particularly outside of the United States, but a .22 caliber pistol with a long barrel (6-12 inches), loaded with dust shot is useful for collecting fast-moving, diurnal lizards which are difficult to noose (Schmidt 1951). Air pistols firing BBs or pellets are also effective, but tend to do more damage to the specimen. Van Denburgh (1922:42) recommended using fine shot in a pistol, small caliber rifle, or smooth bore gun as “the most satisfactory method of procuring reptiles and frogs.”

Shovel. A shovel is a heavy item that can be hard to pack, but it can be very useful for digging trenches, pitfall traps, and extricating field vehicles from tactical miscalculations. The legendary US Army-style folding entrenchment tool is a good compromise, especially if you have an assistant who will do the digging.

Slingshots and Bean Shooters. These have been suggested as alternatives to pistols. Slingshots take some skill to use and a ready supply of ammunition (Engelhardt 1917), but can be very effective.

Snake Handling Devices. A large number of hooks, nooses, clamps, tongs, rakes, and secure buckets have been developed for handling snakes. Several manufacturers make short tongs and hooks that are easy to pack and carry in the field (see *Appendix III*). Do not use collapsible hooks when handling venomous snakes. Snake handling devices include containers (Freed and Freed 1983; Gillingham et al. 1983; Greenbaum 2003); hooks and rakes (Glusenkamp 1995); injecting sticks (Jennings and Vindum 1991); strap and restraining sticks (Gregory et al. 1989; McDonald 1964; Ward and Harrell 1978); tongs (Pillstrom 1954); tubes (King and Duvall 1984; Murphy 1971; Rivas et al. 1995; Sutherland and Hampton 1961; Walczak 1991); tubes modified with a locked end and internal noose (Penner et al. 2008); and wire mesh cable holders (Mauldin and Engeman 1999). Other general references to snake handling devices include Almandarz (1986) and Altimari (1998).

Traps. There are many types of traps and trapping techniques for amphibians and reptiles. Traps must be checked frequently, throughout the day and night, or disabled so that they will not trap unwanted animals. Following is a list of references to traps by type of trap.

Adhesive Traps (Sticky Traps). Commercially available glue boards (intended for catching rodents or insects) can be used, or custom shapes and sizes made by applying glue board adhesive to a sturdy cardboard backing (see *Appendix III*). Live animals may be removed from adhesive traps by using diluted vegetable oil or mineral oil. Adhesive traps must be checked frequently. It is advisable to anchor traps

securely to a fixed object. Remove animals from the traps immediately by gently working a small amount of the diluted oil under the animal with your fingers. For more details on using adhesive traps, refer to Bauer and Sadler (1992); Downes and Borges (1998); Durtsche (1996); Glor et al. (2000); Rodda et al. (1993); Rodda et al. (2005); Vargas et al. (2000); Whiting (1998); Whiting and Alexander (2001); and Zani and Viti (1995).

Aquatic Traps: A simple funnel trap for aquatic larvae can be made out of an empty two-liter plastic bottle by cutting off the top and inserting it backwards into the bottle. In many areas, indigenous hunters weave aquatic funnel traps from plant fibers. General descriptions may be found in Adams et al. (1998); Carr et al. (2014); Graham and Georges (1996); and Richter (1995). Surface aquatic funnels are described by Casazza et al. (2000). Factors that affect efficacy of aquatic funnel traps are discussed by Lauck (2004). Traps for amphibian larvae in seasonal forest ponds may be found in Beuch and Egeland (2002). Klemish et al. (2013) described how to position aquatic traps to minimize accidental death of trapped frogs. Traps have been described for salamanders (Carpenter 1953; Griffiths 1985; Johnson and Barichivich 2004; Smith et al. 2009), hellbenders (Briggler et al. 2013), larval amphibians (Smith and Rettig 1996); snakes (Camper 2005; Fraker 1970); and turtles (Feuer 1980; Gamble 2006; Glorioso and Niemiller 2006; Iverson 1979; Legler 1960a; Mansfield et al. 1998; Selman and Baccigalopi 2012; Sharath and Hegde 2003; Thomas et al. 2008; Tran and Moorhead 2006; Tucker 1994). Singleton et al. (2013) evaluated escape rates of trapped turtles.

Arboreal Funnel Traps. Davis et al. (2008); Fritts et al. (1989); Rodda and Fritts (1992); Savidge (1987); Vogt (1987); and Zani and Vitt (1995).

Bal-chatri Traps. These are traps for basking turtles, made by attaching monofilament slip nooses to a section of poultry mesh fencing that is fitted around a basking site (Braid 1974).

Funnel Traps. These traps may be made from wire mesh, netting, plastic, or metal. A funnel trap consists of a cage or other container with a funnel-shaped entrance (Fig. 9). The funnel opening makes it difficult for an animal to escape the trap once it has entered.

Live Traps. Mammal-style live traps may be used for larger lizards, or the traps may be modified to accommodate a particular species (Doan 1997; Durden et al. 1995; Heatwole et al. 1964; King et al. 1994; Legler 1960b).

Pitfall Traps and Trenches. The simplest form of pitfall trap is merely a hole dug in the ground. I have seen these used for successfully for turtles, baited with a piece of fruit stuck on the end of a stick protruding from the bottom of the trap (Carr et al. 2014). The use of poisons or other killing agents in pitfall traps is not recommended because most of the available chemicals are inhumane, the interval between death and fixation is likely to be unacceptably long, and the wrong species may be killed by accident. For general references on the use of pitfall traps, see Borden (2008); Corn (1994); Karns (1986); and O'Shea (1992). Evaluations of pitfall traps are provided by Banta (1957); Greenberg et al. (1994); Sutton et al. (1999); and Vogt and Hine (1982). Flip-top pitfall traps are described by Christiansen and Vandewalle (2000) and Nadorozny and Barr (1997). Crawford and Kurta (2000) evaluated the



Figure 9. Funnel trap (Henry Fitch Natural History Reservation, University of Kansas).

color of traps. Daoust (1991) discussed ways to avoid the dehydration of trapped animals. Ferguson and Forstner (2006) discussed the exclusion of predators. Whitaker (1967a) discussed using bait in pitfall traps. Perkins and Hunter (2002) found that the use of sticks in pitfall traps reduced the rate of small mammal captures but did not affect amphibian capture rates. Mazerolle (2003) found that rimmed containers were more efficient in pitfall traps for amphibians.

Sheet Traps. This technique involves spreading a plastic or cloth sheet on the ground, constructing an artificial habitat on it to lure the unsuspecting animal, then lifting it off the ground for capture (Allan et al. 2000; Mauldin and Engeman 1999).

Shelter Traps. An artificial shelter can also be used to attract and trap animals. Moulton et al. (1996) described using PVC pipe sections; Strong et al. (1993) suggested plastic or cardboard tubes; Thigpin et al. (2010) described using burlap bands for salamanders; Parker (1971) discussed the influence of trap covers.

Terrestrial Funnel Traps. Clark (1966); Dargan and Stickel (1949); Doan (1997); Enge (1997); Fitch (1951, 1987); Greenberg et al. (1994); Imler (1945); Karns (1986); Keck (1994); Lohofener and Wolf (1984); Mansfield et al. (1998); Mao et al. (2004); Mushet et al. (1997); Row and Blouin-Demers (2006); Vogt and Hine (1982); and Zani and Vitt (1995).

Miscellaneous Traps. Bryan et al. (1991) described a spring loaded turtle trap. Heatwole et al. (1964) used baited snap traps and mammal live traps. Lannon (1962) suspended a dry fly over a pitfall trap. Vogt (1941) described a shelter and pitfall trap. Bryan et al. (1991) and Enge et al. (2012) described gopher tortoise traps. Steen et al. (2010) described modification of box traps for snakes. Rice et al. (2006) described a portable, non-invasive trapping array.

Water Pistol. According to Myers (1956), small lizards can be dislodged from walls and trees with a water pistol, which he described as the “E.R. Dunn method.” The newer pump action hydraulic water pistols (e.g., SuperSoaker) provide more force and a larger fluid reservoir.

Specialized Techniques for Particular Organisms

Amphibians. Artificial pools (Gascon 1994).

Anurans. Collecting bullfrogs by hand (Murphy 1965); locating *Heleophryne* eggs (Visser 1971); extracting anurans from burrows (Heemeyer and Lannoo 2010); arboreal pipe trap for treefrogs (Johnson 2005); use of glow sticks for bait in aquatic funnel traps (Grayson and Roe 2007).

Caecilians. Digging in damp areas is said to be a good way to find caecilians—I once asked Edward H. Taylor his secret for using this method so successfully, and he offered me this advice: “Don’t waste your time digging where you are not going to find anything.” Caecilians may be found in tropical forests, rice paddies, coffee and tea plantations, streams and rivers (Hofer 2000). Gower et al. (2004) compared three sampling methods for two caecilian species (digging, pitfall traps, and encounter surveys).

Salamanders. Newt trapping (Boarder 1969); *Cryptobranchus* collecting (Foster et al. 2008; Soule and Lindberg 1994; Williams et al. 1981); plethodontid collecting (Valentine 1963); sampling stream-dwelling salamanders (Luhning and Young 2006).

Crocodylians. General capture techniques (Chabreck 1963; Hutton 1987; Jones 1965; Murphy and Fendley 1975; Walsh 1987); live traps (Elsey and Trosclair 2004); use of electricity (Joanen and Perry 1971); rope nets and trap boards (Webb and Messel 1977); snap snares (Mazzotti and Brandt 1985).

Amphisbaena. Tipping over termite mounts (Pramuk and Alamillo 2003).

Lizards. *Phrynosoma* (Fair and Henke 1997); carrion-feeding varanids (Smith 2004); collecting *Anolis* by distraction with a moving stick, and collecting terrestrial lizards by corralling with 1.5 m long aluminum sheets (Fitch 1973); collecting *Aspidoscelis* with portable wire mesh corral traps (Fitch 1958). Use of oatmeal bait (Jorgensen and Orton 1961); collecting lizards under bridges (Conant 1951); trap-door box for teiids (Rodgers 1939); luring geckos within reach with a laser pointer (Cole 2004); noose trap for arboreal lizards (Bennett et al. 2001); spotlighting geckos (Whitaker 1967b); sand skinks (Rathor 1970); ambient temperature water in a spray bottle for desert lizards (Estrada-Rodriguez et al. 2004); manual restraint of helodermatid lizards (Poulin and Ivanyi 2003); comparison of finding geckoes using natural cover vs. artificial retreats (Lettink 2007). Trapping lizards in rocky habitats with pitfall traps and live bait (Goodman and Peterson 2005). Small terrestrial lizard trap (Khabibullin and Radygina 2005). Collecting geckos by spotlighting at night from a fixed or moving platform and by digging out rodent and insect burrows, termite hills, and the bases of dead shrubs (Szczerbak and Golubev 1996).

Snakes. Locating snake dens (Jackley 1947); trapping water snakes (Franklin 1947); finding *Charina* (La Rivers 1974); use of trained dogs (Klauber 1997; Stevenson et al. 2010); following monkeys to find *Bitis* (Penner et al. 2008); watching turkeys (Smith 1946); using a forked stick to entangle Boiga (Engeman 1998); baited minnow traps for semiaquatic snakes (Winne 2005); trap for large terrestrial snakes (Burgdorf et al. 2005).

Turtles. Bait (Ernst 1965); basking traps (Lindeman 2014; MacCulloch 1978; Petokas and Alexander 1979); decoys (Mansfield et al. 1998); Fyke nets (double-throated hoop nets) and visual searches (Selman and Baccigalopi 2012); restraint (Galbraith and Brooks 1983); sounding (Carpenter 1955); finding turtles in winter (Bishop and Schoonmacher 1921); freshwater turtles (Bider and Hoek 1971; Fidenci 2005; Iverson 1979, 1991; Kennett 1992; Lagler 1943; Legler 1960a; Vogt 1980; Webb 1961); *Gopherus* trapping (Bryan et al. 1991); *Graptemys* (Chaney and Smith 1950; Wahlquist 1970); softshell turtles (Robinson and Murphy 1975); use of floating pitfall traps and see-saw traps (Sharath and Hegde 2003). Collecting softshell turtles by water-goggling (Webb 1962). Collecting *Malaclemys* by probing open areas in marshes at ebb tide, or by dip netting or drag netting at high tide (Coker 1906). Use of dogs (Cablak and Heaton 2006). Trap modifications to reduce accidental turtle mortality (Bury 2011).

Evaluations of Capture Techniques

General. Mitchell et al. (1993); Parris et al. (1999); Sutton et al. (1999); and Jenkins et al. (2003) compared the effectiveness of funnel traps and pitfall traps used with drift fences. Effect of funnel trap type and pitfall trap size on capture rates (Maritz et al. 2007). Comparison of artificial covers (Hampton 2007).

Drift fences, funnel traps, shelters, and pitfall traps. Arntzen et al. (1995); Banta (1957); Brathwaite (1983); Browne and Hecnar (2005); Bury and Corn (1987); Campbell and Christman (1982); Christiansen and Vandewalle (2000); Enge (2001); Friend et al. (1989); Greenberg et al. (1994); Hampton (2007); Hobbs and James (1999); Jenkins et al. (2003); Lohofener and Wolfe (1984); Maritz et al. (2007); Morton et al. (1988); Olson and Warner (2003); Parmelee and Fitch (1995); Smith and Rettig (1996); Sutton et al. (1999); Vogt and Hine (1982).

Amphibian collecting techniques. Anurans (Brown 1997); salamanders (Briggler et al. 2013; Cooke 1995; Fronzuto and Verrell 2000; Griffiths and Raper 1994; Haan and Desmond 2005). Willson and Dorcas (2003) found funnel trapping for stream salamanders to be more efficient than dipnetting. Pitfall trap designs (Stevens and Paszkowski 2005). Evaluation of commercial aquatic funnel traps (Palis et al. 2007).

Snake collecting techniques. Fitch (1992); comparison of artificial covers and line transects (Olson and Warner 2003).

Turtle collecting techniques. Fidenci (2005); Frazer et al. (1990). Comparison of basking traps and baited hoop traps (Browne and Hecnar 2005).

PHOTOGRAPHY

With the availability of small, easy-to-use digital cameras, there is no excuse for not photographing habitats and specimens in the field. A wide variety of digital cameras are available of varying sizes and levels of sophistication. Minimally, a digital camera for field use should be rugged, have a macro feature capable of filling the frame with a small animal, be capable of taking high resolution images, have a flash, and have a large memory card. The type of camera you choose to carry will depend on the quality of images you want to bring back. Images suitable for identification of specimens and use online can be made with a small, relatively inexpensive self-contained cameras; quality images for publication will require a more complex camera and lens combination. If very high quality images are desired, check the “About Our Cover” section of recent issues of *Herpetological Review* for details of the equipment used to take the photographs used on the journal cover.

Before you go in the field, practice using your camera on amphibians and reptiles or similar-size objects, as well as taking habitat shots, so that you become familiar with the camera functions. For use on extended field trips, carry replacement batteries and extra memory cards. Keep the camera in a case when it is not in use, and carry a lens-cleaning cloth with you.

When photographing an animal, pose it on a natural background for context. Werner (2011) recommends photographing specimens in their natural habitat both with and without size and color scales in the frame, and photographing animals on mirrors to show both dorsal and ventral patterns. Record the numbers of the images of each specimen photographed in the field catalog.

Reptiles and amphibians usually have to be placed in position several times before they will remain still long enough to be photographed. Be patient. Amphibian and reptile photography is best done with two people—one to take the photograph, the other to wrangle the animal. One trick that will save a lot of frustration when posing animals is to position the specimen where you want it on the background, then cover it with an object or your hand (Twiest 1968). Compose your shot, remove the covering, and snap the picture. Specimens may be placed in a bucket of water and allowed to swim for a time to tire them sufficiently that they can be more easily posed (J. Caldwell, pers. comm.). Some photographers prefer to chill specimens in a refrigerator or ice chest before posing them, but this technique may alter the colors of the animal. Kaiser and Green (2001) have suggested rubbing the venter of small animals with a small amount of benzocaine (ethyl p-aminobenzoate) based oral analgesic (e.g., Oragel) as an anesthetic. This should be done with caution, as the same chemical is also used to euthanize animals. See Mobbs (1978) and Twiest (1972) for other hints.

AUDIO RECORDING

The first known recording of an animal sound was a bird that was recorded in 1889 (Boswall 1969). Nevertheless, the recording of frog calls is still ignored by most field herpetologists, despite the fact that there are species that can only be easily identified by their calls. With the advent of lightweight digital

recorders, it has become much easier to make quality recordings with high research or documentary value. Differences in vocalizations are an important way to distinguish between some closely related species of frogs, and many frogs and some lizards utter sufficiently distinctive calls that a recording can serve as a voucher for the presence of the species, without the need to collect a specimen. For more information on field recording, refer to Heyer (1994).

In the past, getting decent quality recordings of frog calls required taking expensive and often heavy tape recording equipment into the field; modern digital recording equipment is much lighter in weight and easier to use. Reviews and discussions of recording equipment for making high quality field recordings in the field may be found on the websites for the Cornell Laboratory of Ornithology's Macaulay Library and the Wildlife Sound Recording Society (see *Appendix III*).

Recorder. Although it is possible to record frog calls in the field using a cell phone or small voice recorder, the quality of these recordings is insufficient for research use, although they can be used for identification purposes. Both cell phones and voice recorders are designed to record the limited frequency range used by the human voice, rather than the full range of frequencies likely to be emitted by singing anurans.

A recorder for field use should be durable enough for use outdoors under the conditions in which frogs are likely to be recorded (which includes cold and damp weather), and be both lightweight and easy to carry. Select a recorder with a sampling rate of at least 48kHz with a 24 bit depth, and a good microphone preamp. The recorder should have the capacity to save recorded sounds in an uncompressed format (e.g., as wav or aiff files); compressed files (such as mp3 files) will undergo some frequency loss during compression. Digital recorders should have a large memory storage capacity and use SD cards or have a USB port to simplify access to the recordings. The recorder should have a recording level that can be manually controlled, with a recording level meter that can be read in low light. Make sure that the recorder you select has input jacks that accept the microphone you chose, and a headphone jack for playback. An external playback speaker is also useful, particularly for stimulating frogs to call. A good recorder for field use should have batteries that can be easily replaced or recharged, and a sturdy carrying case.

Microphone. It is the microphone that determines which sounds and what quality sounds the recorder will have available to record. Select a microphone with wide dynamic range that is durable enough for field use. Most frogs can be approached closely without having to resort to a parabolic reflector, although both parabolic reflectors and shotgun microphones can be very useful to obtain high quality recordings in the field.

Field Recording Techniques

Before heading off to the wilds, don't just know how to operate your recording equipment, know how to operate it in the dark. Sinking crotch deep into a leech-infested swamp with the recorder hanging around your neck and the batteries in your headlamp giving out is no time to discover that you can't remember which button is record and which is playback.

Have the following ready when you want to record frog songs:

1. A notepad to jot down data which you might accidentally erase from the recording, and to help you keep the recorded segment numbers straight.
2. A series of sequentially numbered water-resistant tags to drop into the plastic bag with the individual specimens you record and collect.
3. Enough plastic bags to keep each recorded individual separately.
4. A thermometer to determine air and water temperature, or a digital combination thermometer and relative humidity gauge. This information may be crucial for the later interpretation of the recording. Be sure to position the device away from your body when taking measurements.

Before you began recording an individual frog, make a recording of the entire chorus (if present) and background noises at the site for a few minutes. This will be useful later to help identify and filter out extraneous noise, and may also be useful to encourage the frogs to call again once you have got your recorder all ready and they suddenly go silent (Legler 1964). Using a red light, or covering your headlamp with red cellophane or clear red plastic will enable you to see the recorder while causing less disturbance to the subject you are trying to record.

Whether recording a chorus or an individual, follow these basic steps:

1. Position yourself in a place where you can stand still and get a clear recording of the desired sound.
2. Using the recording level meter and monitoring system, check to confirm that you are picking up the sound you want to record.
3. Give the individual frog time to begin behaving normally after you are in place to record. Recording diurnal frogs requires extra care and patience, as they are much more wary than nocturnal species. Locate the frog you wish to record, set up the equipment, then wait patiently for the frog to resume calling.
4. Start the recording, holding the microphone as still as possible and shielded from the wind. Record the frog for at least five minutes without interruption. This length of recording is necessary for analysis of calls per minute, intervals between calls, and to record reciprocal calls from other males.
5. When the desired sequence has been recorded, collect the individual you have recorded. Recordings are much more valuable when you collect the calling frog as a voucher specimen. Bag each recorded individual separately with a number tag in the bag.
6. Play the recording back to make sure you got what you intended to record. At the end of the segment, record the segment number, date, locality, time, air and water temperature, relative humidity, species recorded, and identifiable background noises (including other species of frogs).
7. If previously recorded calls are used to stimulate a chorus or individual to begin calling again after they have been disturbed (Legler 1964), note this in the audio log in your field book along with other information about the recording.

SNAKEBITE

Although venomous snakebite is not a common occurrence among field workers, it does happen. The more often venomous snakes are handled, the greater the risk from snakebite. Always presume that a snake is venomous if you are not positively sure of what it is. Always handle snakes by the safest method available—for example, use tongs or tubes instead of pinning the snake and picking it up by hand (Altimari 1998).

If snakebite does occur, check for signs of envenomation before beginning treatments such as antivenin or surgery. It is estimated that at least 30–50% of venomous snakebites are dry bites, in which no envenomation occurs (Norris and Minton 1995). Be aware that elapid bites may not show symptoms until several hours after the bite.

Do not use ice, electric shock, tourniquets, “cut and suck” treatment methods, or topical agents to treat snakebite, as these may increase the risk of tissue damage or death.

Viper bites. Signs and symptoms from viper bites may include pain, swelling, bloody ooze from the puncture marks, weakness, dizziness, nausea, vomiting, chills, sweating, odd taste sensations, headache, tingling of the skin, and muscle weakness.

- If you are within a few hours of medical care, get to a qualified physician as soon as possible.
- If you are more than a few hours away from medical care, a pressure bandage (not a tourniquet) may be applied, wrapping the entire bitten appendage. Apply with the same pressure as for a sprained ankle.
- Although some people recommend the use of an Extractor snakebite kit (see *Appendix III*) immediately after the bite, there is evidence that this device may not remove a significant amount of venom and may cause more harm than good. Its use will also interfere with the application of a pressure bandage. In any case, do not make any cuts in the skin.
- Support the bitten area with a splint or sling.
- Transport the victim to medical help as soon as possible.
- The victim may be given fluids (if there is no nausea), but not beverages containing alcohol or caffeine.
- At the hospital, ask the attending physician to start an intravenous (IV) line immediately and to consult with the regional poison control center.
- Before antivenin is administered, remind the physician to be prepared for anaphylaxis (an allergic reaction to the antivenin).

Elapid and hydrophiid bites. Signs and symptoms from elapid and hydrophiid bites may include pain, numbness, muscle weakness, paralysis, difficulty breathing, difficulty swallowing, and difficulty speaking.

- Apply a pressure bandage (not a tourniquet) as quickly as possible, wrapping the entire bitten appendage. Apply the pressure bandage with the same pressure as for a sprained ankle.
- Do not use an Extractor or any other type of snakebite kit. Do not make any cuts in the skin.
- Support the bitten area with a splint or sling.
- Transport the victim to medical help as soon as possible.

- The victim may be given fluids (if there is no nausea), but not beverages containing alcohol or caffeine.
- At the hospital, ask the attending physician to start an intravenous (IV) line immediately and to consult with the regional poison control center.
- Before antivenin is administered, remind the physician to be prepared for anaphylaxis (an allergic reaction to the antivenin).

PART III. PRESERVATION OF SPECIMENS

Grave Digger: ... A tanner will last you nine year.

Hamlet: Why he more than another'?

Grave Digger: Why, sir, his hide is so tanned with his trade that he will keep out water a great while, and your water is a sore decayer of your whoreson dead body.

—William Shakespeare, *Hamlet*, Act V, Scene I

The sooner that amphibians and reptiles are processed after they are collected, the better quality the resulting specimens will be. Some field workers prefer to preserve specimens immediately after capture, including those collected at night, which means working by artificial light; other collectors wait until morning to take advantage of natural light. An advantage to preserving specimens immediately after capture is that stomach contents will be better preserved.

Field notes should document critical preservation information that may be important for future use of the specimens. For each collecting trip, include a description of the methods used for killing and preserving, a list of chemicals and their sources, and how the chemicals were mixed (e.g., formaldehyde purchased locally and diluted with stream water). Note any variations in materials or procedures in the individual specimen accounts as necessary. In the field catalog, note the time of capture and the time of death and preservation of individual specimens.

The amount of care taken with the initial preservation of a specimen is arguably the most important factor in determining how useful the specimen will be in a collection (see *Conservation of Herpetological Collections*). Follow proper procedures, and make sure that chemicals used for fixation and preservation are fresh and of proper strength.

IACUC COMMITTEES

Most universities and other research institutions require field practices to be approved by an Institutional Animal Care and Use Committee (IACUC), as required by federal law. The IACUC system originated with the Laboratory Animal Welfare Act of 1966 (Public Law 89-544), which was passed in response to a series of alarming reports in the press concerning cruel treatment of laboratory animals and the lack of nationwide standards for animal care (Rozmiarek 2014). Further public pressure from various animal welfare organizations (as well as a number of break-ins and vandalism incidents at animal research facilities across the country) resulted in the passing of Public Law 99-198, the Food Security Act, in 1985; Subtitle F—Animal Welfare (also known as the Improved Standards for Laboratory Animals) of this act defined humane care and required the establishment of regulations for animal housing, required minimization of pain and distress to laboratory animals, and established the role of the IACUC at institutions receiving federal funding (Rozmiarek 2014). The IACUC regulations took effect on 30 October 1989 following the publication of the final rules for Parts 1 and 2 of the Animal Welfare Act in the Federal Register. The final rules for the Standards portion of this act were published on 15 February 1991 (9 CFR

Part 3). A number of amendments and clarifying regulations have been added to the Animal Welfare Act in the intervening years.

Details concerning under what circumstances an IACUC is mandated, as well as references to the relevant legislation and regulations, can be found in Brown and Shepherd (2014a, 2014b). In short, the IACUC is required to review and enforce the animal care and use programs and regulations at its institution every six months, investigate complaints, and perform regular inspections of animal facilities “including animal study areas and satellite facilities” (Brown and Shepherd 2014a:13). The vast majority of the activities regulated under IACUC legislation concern use of laboratory animals; however, at most institutions, field work also comes under the jurisdiction of the IACUC. Federal regulations require that the IACUC be composed of “at least one veterinarian, one practicing scientist experienced in research involving animals, one member whose primary concerns are in a nonscientific area, and one individual who is not affiliated with the institution in any way and is not a member of the immediate family of a person who is affiliated with the institution” (Brown and Shepherd 2014b:25-26). It is not uncommon for field researchers to find that the IACUC members at their institution are unfamiliar with and therefore do not understand fieldwork. Should this happen, the researcher should (1) become familiar with relevant IACUC legislation and interpretation; (2) make sure that the IACUC members have access to the proper guidelines (discussed below); and (3) insist that someone who regularly does field work involving the collecting, handling, and euthanizing of amphibians and reptiles be appointed as a member of the IACUC.

Institutional Animal Care and Use committees generally follow the guidelines provided by the American Veterinary Medical Association (AVMA) although the guidelines are not legal standards (Danneman 2014). It is important to keep in mind when dealing with an IACUC that neither Public Health Service (PHS) policy nor the Animal Welfare Act Regulations (AWAR) address the use of amphibians or reptiles in detail (Huerkamp et al. 2014), leaving regulation up to the “professional judgment” of members of the IACUC (Huerkamp et al. 2014:256). Furthermore, expert opinion recommends that each IACUC follow the “recommendations for specific species... often available from organizations that have an interest in the appropriate care and use of these species in the laboratory and field studies” (Danneman 2014:256). For amphibians and reptiles, expert recommendations are available in Breaupre et al. (2004); Pough (1992); and Schaeffer et al. (1992). The most significant guidelines for field work with amphibians and reptiles are those established by the American Society for Ichthyology and Herpetology (ASIH) in “Guidelines for use of live amphibians and reptiles in field and laboratory research,” which is available on the ASIH website (see *Appendix III*). The ASIH guidelines address the role of the IACUC, collecting and acquisition, sampling, and euthanasia practices, among other topics. It is recommended that field work proposals submitted to an IACUC be accompanied by copies of the relevant amphibian and reptile specific publications, as this literature is likely to be unfamiliar to most IACUC members.

THE FIELD KIT

Descriptions of a variety of field kits to carry collecting and preserving gear have been proposed, ranging from a custom box (Maslin and Swenson 1971) to a small tackle box (J. Campbell, pers. comm.). Whatever the size and shape, a field kit should contain the following (see also *Appendix II*):

1. Hardening trays (polypropylene) with tight fitting lids (the first time you spend the night in a room with a tray full of formaldehyde and a leaky lid, you'll know why...).
2. Paper towels to line the hardening trays.
3. Formaldehyde (full strength).
4. Packets of pre-mixed buffers for formaldehyde.
5. Small plastic graduated cylinder or beaker to measure formaldehyde and water when mixing.
6. Container with tight fitting lid for mixed 10% neutral buffered formalin solution.
7. Field tags, sequentially numbered, with strings attached, ready for affixing to specimens.
8. Field book with 100% rag or spun-bonded polyethylene pages.
9. A technical pen with a fine point.
10. A good supply of archivally stable ink (see *Field Notes*).
11. A number two pencil, pencil sharpener, and good quality eraser.
12. A few containers for holding live specimens during processing.
13. Bottle forceps for handling preserved specimens.
14. Chemical agents for euthanizing specimens.
15. Small forceps, probes, and dissecting needles for positioning specimens in the hardening tray.
16. A variety of syringes and needles for administering euthanizing chemicals and injecting fixative into specimens.
17. A teaspoon or small tea strainer for handling aquatic larvae.
18. Scissors—small, sharp scissors for cutting specimens; bandage scissors for cutting tags, paper, and cheesecloth.
19. Scalpel handle and a box of new blades.
20. Skinning knife (for preparing large specimens).
21. Separate set of dissecting tools, kept very clean, for taking tissue samples (scissors, forceps, probe, scalpel handle and blades); soap and 10% bleach solution for cleaning tools after taking each tissue sample.
22. Thread for sewing parts back on specimens.
23. Extra tag string for restringing tags (in case the wrong ones are tied on a specimen).
24. A hand lens (10x) for specimen identification.
25. A color chart with metric size scale.
26. Scales (spring or digital).
27. Chemical splash goggles that meet ANSI Z87.1-1989 standards.
28. Cheesecloth and plastic bags for packing specimens.
29. Neoprene gloves for handling specimens in formaldehyde.
30. Variety of sewing needles for attaching tags to limbless animals and sewing specimens up when necessary.
31. Large containers with good lids to hold specimens in fluid after tagging.

FIELD NOTES

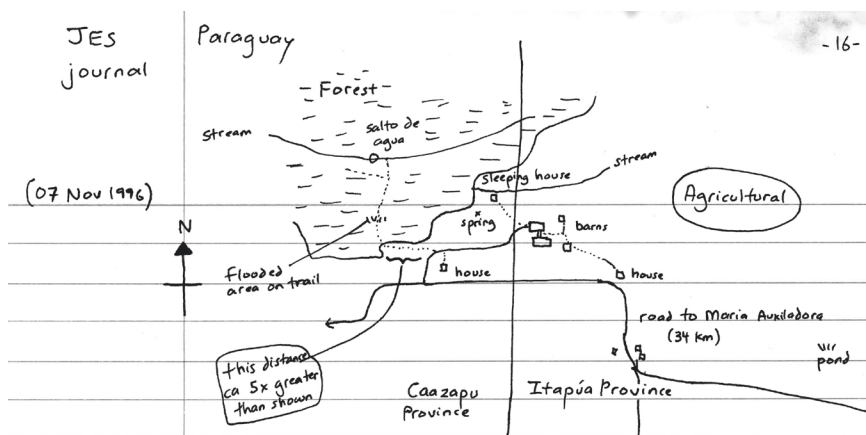
Field notes are the critical documentation that give validity to scientific specimens. Writing good field notes requires patience, practice, and diligence. The standard for natural history field notes was set by Joseph Grinnell at the Museum of Vertebrate Zoology at the University of California in Berkeley. Grinnell began writing field notes in 1908, and made his last field note entry just five days before his death in 1939. Grinnell's techniques and methods have been described and amplified by Herman (1986). An alternative based on Grinnell's system may be found in Hall (1962). An extensive review of the history, styles, and uses of field notes, as well as numerous suggestions for making notes and field sketching, can be found in *Field Notes on Science and Nature* (Canfield 2011). Over time, most collectors develop their own particular style for field notes that meets their research and specimen documentation needs—what is critical is that the notes be written so that they can be easily read and unambiguously understood by other people using the preserved specimens long after the collector is gone. Writing good field notes takes practice. I recommend always keeping field notes when traveling, whether collecting or not, as a good way to perfect your technique and to keep in practice. Field notes must be clear and legible, hence printing is preferred over cursive writing.

Field notes are extremely valuable, both for your present purposes and for future reference. Treat your field notebook carefully, and take it with you as carry-on when traveling by airplane. Deposit the originals of your notes in a reliable museum archive, preferably along with your specimens. Never rely on recorded audio notes or a computer for recording field notes, as data in either of these formats may be easily lost—a digital backup copy of the notes and catalog information is useful, but do not rely on electronic records as your sole source of notes. No recording medium is as permanent as archival quality ink and paper (Simmons 1998). Use acid-free, 100% rag paper (or spun-bonded polyethylene sheets) for both field journal and field catalog. I recommend using a technical pen with Higgins Black Magic or equivalent carbon-based ink. Although some disposable pens contain ink rated as archival, most do not (most disposable pens contain pigments other than carbon black and other ingredients that affect the permanence of the ink such as surfactants, waxes, and solvents). If you must use disposable pens, purchase them from a reliable archival supply house (see *Conservation Supplies* in *Appendix III*) and test them on your field book paper first (a recommended disposable pen is the Sakura Pigma Graphic Pen). Never use colored inks for field notes as colors other than black are rarely archival.

Field notes consist of two separate but related documents—the field journal (Fig. 10) and the field catalog (Fig. 11), which are discussed separately below.

Field Journal

The field journal is a narrative account that includes a detailed trip itinerary, descriptions of habitat and weather (including data on rainfall, temperature, sunlight, and moonlight), observations on species occurrence and behavior, and descriptions of collecting and preparation techniques. It is important to use a format that organizes the information in a clear, logical arrangement. Herman (1986) recommended a good standard format; a modified version of



Sky cleared in afternoon. No moon. Worked the seepage area by the spring, across the creek from the sleeping house -- nothing calling. Walked forest trail to salto de agua, found only one Leptodactylus at the flooded area. Forest vegetation was dripping, leaf litter wet. No sign of herps other than the one frog collected and another calling at the flooded area.

08 Nov 1996 08 November 1996. Friday. Day dawned clear and sunny; became overcast between 9:00-9:30 AM. Was 22.5° at 8:45 a.m. Went hiking along the road in from Maria Auxiliadora, searching for other areas to collect. After 1.5 hours of walking, located a cattle pond and a path that takes about 25-30 min. walking to reach it. Returned after dinner to find anuran activity was high, the chorus of Hyla faber deafening (though they were very difficult to approach in the water). Also found calling Hyla nana in abundance, some unknown metamorphosing hylids, two Scinax fuscovaria, calling Leptodactylus ocellatus (found only one male, but three females), and around the edge on the dam end of the pond, Phyllaemus biligonigerus. The pond was difficult to work. The bottom was soft and mushy, much of the bank was thorny second-growth vegetation. One Hyla faber got me with his prepollical spine under the fingernail.

Figure 10. Field journal.

28 July 1986

- 6 -

28 July 1986

Ecuador: Esmeraldas: Borbón, Campamento Forestal Borbón, 30 m

1465 Bufo marinus

J.E. Simmons. On lawn in front of camp headquarters at night.

1466 Eleutherodactylus cf. achitinus

J.E. Simmons. Forest floor leaf litter by day. Dorsum dark tan, deep brown stripe from nostril to behind tympanum, faint yellow in groin, chin & venter white, faintly mottled grey

1467 Gonatodes caudiscutatus

M.T. Neilson. On wall of building by day. Dark grey dorsum, grey-brown venter. Some tiny bronze flecks on dorsum.

1468 Eleutherodactylus achitinus

J.L. Carr. Low vegetation at night. Like JES 1466

1469 Eleutherodactylus cf. achitinus

M.T. Neilson. Low vegetation at night. Like JES 1466, only dorsum darker, yellow extends to venter, orange in groin.

1470-71 Eleutherodactylus

J.L. Carr. Low vegetation at night. Dorsum brown with darker brown markings. Black stripe from nostril to behind tympanum. Tan bars on sides, limbs. Venter and chin white with grey mottling, underside of thighs & lower leg faint orange. #1471 without orange (much smaller specimen).

Figure 11. Field catalog.

that format that I use is shown in Fig. 10. Each page of the field journal should be headed with the date and locality.

Field Catalog

The field catalog is a list of specimens collected and their field numbers, with detailed descriptions and collecting information. Herman (1986) recommended a good standard format; a modified version of that format that I use is shown in Fig. 11. Each page of the field catalog should be headed with the date and the complete collecting locality, including GPS data. For each entry, record the field number and scientific name of each specimen. Use a single, sequential set of field numbers throughout your collecting career—never re-use field numbers. List all collectors by name or initials (if initials are used, include a list of collector names as well). Next, describe where the animal was found (microhabitat) and what it was doing. Describe the specimen, paying particular attention to details that may be lost or modified by preservation, such as live weight and colors. Use a standard reference color chart. Hess (1988) and Zucker (1988) both recommend the Munsell System; Tucker et al. (1991) provides a comparison of several color charts. Use good quality spring scales or portable electronic scales for determining weight (calibrate the scales before taking them into the field). Include references to any photographs, recordings, tissue samples or other data related to the specimen.

PROCEDURE FOR PROCESSING SPECIMENS IN THE FIELD

The goal of preservation is to prepare good specimens for research use, record critical data, and obtain necessary photographs and tissue samples while keeping the interval between specimen death and preservation as short as possible. To achieve this goal requires organization of the work area and advance planning (Fig. 12). The recommended basic field procedure is the following:

1. Select as comfortable a work area as possible, and arrange the preserving gear, field catalog, photographic equipment, tissue sampling gear, and specimens to be processed. Make sure you have the necessary data from other collectors to make the complete catalog entry for each specimen.
2. Prepare the chemicals needed (e.g., 1:9 solution of buffered formaldehyde) before starting.
3. Arrange the specimens in the proper order for cataloging. This order may be based on collecting site, species, time of collecting, collector, or several of these factors.
4. Take photographs of the specimens before they are euthanized.
5. Select those specimens you wish to take blood or tissue samples from before you began euthanizing specimens, as these will require special handling (see *Blood and Tissue Samples*).
6. Euthanize and process the specimens individually, recording data for each individual in the field catalog as you go (see *Field Catalog*).
7. Lay out the specimens in the hardening trays in the order in which they are cataloged (to facilitate tagging later), or tag the specimens prior to positioning them in the hardening tray.
8. Always clean up any chemical spills promptly.
9. Clean and put away all preserving equipment and rinse out collecting bags at the end of each preservation session.



Figure 12. Jennifer Pramuk and Hugo Alamillo preserving specimens (17 km E of Karonay, Itapúa, Paraguay).

10. Write your field journal (see *Field Journal*) immediately after the specimens are preserved.

Killing

Specimens should be dispatched quickly by a method that will cause minimal pain and leave the specimen relaxed (Anon. 1987). Most field workers based in the United States will be required to use procedures in compliance their Institutional Animal Care and Use Committee (see *IACUC Committees*).

Reptiles

The preferred method of killing reptiles is by injection of aqueous sodium pentobarbital (e.g., Nembutal, Fatal Plus) into the heart (Karlstrom and Cook 1955; Lowe 1956). Sodium pentobarbital is a controlled substance—check with a legal supplier of this chemical (e.g., a pharmacy or chemical supply company) for permitting information. Use approximately 1.0 to 5.0 cc for large specimens, less than 1.0 cc for smaller specimens (Lowe 1956). The solution may be diluted 1:5 to 1:10 with water, depending on the size of the specimen. The heart may be located in most reptiles by gentle pressure from one or two fingers (not your thumb) on the chest. As a general rule, in lizards the heart is just below the junction of the arms in the middle of the chest; in most snakes, the heart is about 1/4 of the way down the body from the head. Sodium pentobarbital may also be injected into the brain via the upper inside surface of one of the nares (L. Vitt, pers. comm.).

Other Methods for Killing Reptiles

- Injection of MS 222 (also called tricaine, tricaine methanesulfonate, or Finequel) (Karlstrom and Cook 1955; O'Shea 1992). Use about ½

teaspoon in 20 ml of water to make an injectable solution (L. Grismer, pers. comm.). Use a freshly mixed solution as it loses effectiveness after a few days.

- Injection of lidocaine [also known as lidocaine hydrochloride, 2-diethylamino-N(2,6-dimethylphenyl) acetamide, Anestacon, Dilocaine, L-Caine, Lida-Mantle, Nervocaine, Octocaine, or Xylocaine] to the heart (Páez et al. 2002) or into the brain via the upper inside surface of one of the nares (L. Vitt, pers. comm.).
- Livezey (1958) recommended the use of procaine hydrochloride for reptiles and amphibians. Other names for this drug include procainamide hydrochloride, Procan, Procanbid, and Pronestyl. A 10% mixture is prepared by dissolving tablets (which contain 0.07 g of procaine hydrochloride) or crystals in water, or by obtaining the drug as a prepared solution. About 0.05 cc per gram of body weight should be injected into the pleuroperitoneal cavity just posterior to the heart.
- Lambert (1967) suggested using Anectine (succinylcholine chloride). Other names for this drug include diacetylcholine chloride, Quelicin, and Sucostrin. It is available as a white powder. The recommended solution is 25 mg/cc of water. Inject into the abdominal and pericardial cavities. Small reptiles require 0.05 to 0.20 cc; large reptiles require up to 2.5 cc or more.
- Reptiles may be euthanized by placing them in a killing jar containing cotton soaked in nail polish remover (ethyl acetate; see O'Shea 1992), or acetone (L. Grismer, pers. comm.).
- Application of a small amount of benzocaine (ethyl p-aminobenzoate) based oral analgesic (e.g., Oragel) to the head (Altig 1980) or venter (Chen and Combs 1999) may be used to kill small, thin-skinned reptiles.
- Chloroform.
- Ether.
- Less desirable methods of killing include the injection of isopropyl alcohol into the brain through the optic foramen, or by freezing (Meylan 1989). The drawbacks to these two methods are that the cranium of the specimen will be damaged by the former, and the latter may result in a frozen specimen that will not preserve well, due to ruptured cells and the inability of fixatives to penetrate frozen tissues (see *Frozen Specimens*).

Amphibians

The preferred method for killing amphibians is to submerge them in a solution of hydrous chlorobutanol (1,1,1-trichloro-2-methyl-2-propanol; also known as Chloretone; see Snyder 1915). Prepare a saturated solution by dissolving hydrous chlorobutanol crystals in 95% ethyl alcohol; use a few milliliters of the saturated solution in about 500 ml of water. Alternately, a killing solution may be prepared by dissolving one teaspoon of hydrous chlorobutanol crystals in one liter of water (McDiarmid 1994).

Other Methods for Killing Amphibians

- Injection of or immersion in MS 222 (also called tricaine, tricaine methanesulfonate, or Finequel) (Cecala et al. 2007; Karlstrom and Cook 1955; O'Shea 1992; Peterman and Semlitsch 2006; Schultz and Dawson 2003). Use about ½ teaspoon in 20 ml of water to make an injectable

solution (L. Grismer, pers. comm.). Use a freshly mixed solution as it loses effectiveness after a few days.

- Application of a small amount of benzocaine (ethyl p-aminobenzoate) based oral analgesic (e.g., Oragel) to the head or venter (Altig 1980; Brown et al. 2004; Cecala et al. 2007; Chen and Combs 1999).
- Submersion in a 25% ethyl alcohol and water solution (Páez et al. 2002).
- Submersion in a solution of procaine hydrochloride (Livezey 1958). Other names include procainamide hydrochloride, Procan, Procanbid, and Pronestyl. A 10% mixture is prepared by dissolving tablets (which contain 0.07 g of procaine hydrochloride) or crystals in water, or by obtaining the drug as a prepared solution.
- Use of a killing jar containing cotton soaked in nail polish remover (ethyl acetate; see O'Shea 1992).
- Very large amphibians, particularly those with thick skins, may be killed with an injection of sodium pentobarbital or any other of the methods described above for reptiles.

Blood and Tissue Samples

Although a few researchers have claimed some success in extracting DNA from formaldehyde-fixed specimens preserved in ethanol (e.g., Chatigny 2000; see review in Gamble 2014), it is critically important for collectors to prepare fresh blood or tissue samples expressly for this purpose at the time specimens are processed. Failure to prepare tissue samples for molecular studies results in specimens that are less valuable for research purposes.

Non-fatal techniques for collecting blood or tissue samples from live amphibians and reptiles are described in the references listed below, with the caveat that a DNA sample vouchered by a well-preserved specimen is significantly more valuable than just a DNA sample alone.

Tissue. Amphibian buccal swabs (Gamble 2014; Goldberg et al. 2013; Tanaka-Ueno et al. 2006); amphibian toe clips (Gamble 2014; Gonser and Collura 1996); anuran skin swabs (Pichlmüller et al. 2013; Streicher et al. 2014); autonomized tails (Cameron et al. 1998; Cordero et al. 1998; Gamble 2014); carapace notching (Dawes et al. 2008; Mockford et al. 1999); caudal muscle plugs (Haskell and Pokras 1994); claw clips (Lutterschmidt et al. 2010); cloacal swabs (Gamble 2014); feces and fecal sacs (Katz et al. 2013; Tanaka-Ueno et al. 2006); shed skins (Bricker et al. 1996; Clark 1998; Gamble 2014; Tanaka-Ueno et al. 2006); use of commercial filter paper products and epithelial cells (Davis et al. 2002). For a review of sampling techniques specifically for amphibians and reptiles, see Gamble (2014).

Blood. Techniques have been published for gilled amphibians (Rivera and Davis 2013); lizards (Esra 1975); and turtles (Bulté et al. 2006; da Silva et al. 2012). Specific techniques include cardiac puncture in frogs (Rexer-Huber et al. 2012), turtles (Maxwell 1979), and snakes (Branch 1973; Sooter 1955); caudal puncture in snakes (Reinert and Bushar 1991) and turtles (Powell and Knesel 1992; Stephens and Creekmore 1983); coccygeal veins (Haskell and Pokras 1994); neck puncture in turtles (Wibbels et al. 1998); and taking blood from the cervical sinus of turtles (Bennett 1986). Berish and Miller (2012) described a device for restraining turtles during blood extraction. For a review see Gamble (2014).

Tissue Samples from Euthanized Animals. Detailed instructions for documenting, sampling, and preserving tissues for genetic study can be found in Gamble (2014). In brief, tissue samples should be taken as soon as the specimen is euthanized and the sample should be preserved immediately, preferably by flash freezing in liquid nitrogen. If nitrogen is not available, tissues may be preserved in 95% ethyl alcohol (see extensive discussion in Gamble 2014). Samples about the size of the eraser on a pencil should be taken of liver or skeletal muscle. Tools used to take tissue samples should be cleaned with soap and water immediately after use, and then dipped in 10% bleach to prevent contamination of future samples. Gamble (2014) discusses details of lethal and non-lethal tissue removal, labeling, alternative means of preservation, provides detailed procedures to follow to avoid contamination, and discusses long-term preservation of tissues. Exposure to heat, light (particularly ultraviolet radiation), moisture (including high relative humidity), and harsh chemicals may degrade the quality of tissue samples. Before removing tissue from specimens, make sure that your permits allow you to take tissue samples. Although most jurisdictions consider tissues to be the same as specimens, in some areas, special permission is necessary to take tissues for DNA extraction.

Fixation and Preservation

Most specimens of amphibians and reptiles are preserved in fluid. Historically, specimens were cut open and placed in containers of 70% ethyl alcohol for preservation. The two-step process of formaldehyde fixation followed by alcohol preservation was not developed until the late 1890s and early 1900's, after the discovery of the fixative properties of aqueous formaldehyde, and did not become common as a field technique until well into the early 1900s (Ford and Simmons 1997; Simmons 2014). There is a widespread belief among herpetologists that formaldehyde fixation is necessary for the successful preservation of specimens, but the oldest fluid-preserved herpetological specimens in museums were not fixed in formaldehyde. We know that alcohol causes tissues to shrink and colors to fade (due to the loss of pigments and to structural changes), but we also know that formaldehyde fixation disrupts protein chains, making it very difficult (usually impossible) to extract usable DNA from the tissues, causes specimens to darken, and over time, may decalcify bone (Simmons 2014). Specimens may be preserved directly in alcohol, without an aldehyde-based fixative, by using 70% ethyl alcohol, making sure that the alcohol penetrates deeply into the specimens by injecting it or making cuts into the tissues, and changing the alcohol after 24 hr (this step is necessary because the alcohol becomes diluted as it extracts water from tissues).

Fixation is a chemical process that (1) prevents autolysis (the degradation of proteins into amino acids) by the formation of covalent bonds known as crosslinks; (2) coagulates cell contents into insoluble substances; and (3) protects the specimens from microbial deterioration (Simmons 2014; Stoddard 1989). A preservative does not form crosslinks but disorders proteins by altering patterns of hydrogen bonding. Preservatives have been in use for fluid specimens for about 350 years, fixatives for about 100 years.

A good fixative is slightly acidic and penetrates tissues rapidly. The most widely used fixative for herpetological specimens is formaldehyde.

Formaldehyde is sold as an aqueous solution of 37% v/v (or 40% w/v) formaldehyde with a small amount of methanol (6-15%) added to the solution to prevent polymerization. The term *formalin* usually refers to this aqueous mixture of 37% commercial formaldehyde (formalin is an old trade name for commercially produced formaldehyde). The traditional “10% formalin” used to preserve reptiles and amphibians is made by diluting one part commercial formaldehyde with nine parts water, thus “10% formalin” is actually 3.7% formaldehyde in aqueous solution.

When diluted with water, formaldehyde solutions are acidic, in the range of pH 3.0 to 4.6, due to the presence of small amounts of formic acid, either as an impurity remaining from manufacture, or as a result of the oxidation of part of the formaldehyde. Formaldehyde reacts with oxygen in the air to oxidize into formic acid, so it has to be neutralized or buffered. The use of unbuffered formaldehyde will shorten the useful life of a specimen and may cause problems with later attempts to prepare the specimen for other types of study (Simmons 2014; Quay 1974). The pH range achieved by buffering is critical—decalcification may begin at pH 6.4 and below; but the clearing of tissue may start at pH 7.0 and above.

The traditional buffers for formaldehyde were borax and calcium carbonate, but neither of these should be used as they are not stable for very long (Hughes and Cosgrove 1990; see discussion in Simmons 2014). The preferred buffer for formaldehyde is that proposed by Quay (1974): use 4.0 g of monobasic sodium phosphate monohydrate ($\text{NaH}_2\text{PO}_4\cdot\text{H}_2\text{O}$) with 6.5 g of dibasic sodium phosphate anhydrous (Na_2HPO_4) per 1 liter of solution consisting of one part commercial formaldehyde (37%) and nine parts of distilled or deionized water. The two salts can be pre-measured and sealed in small plastic packets, ready for use in the field. When mixing fixative solutions, always measure the water and formaldehyde carefully. Do not try to mix solutions by attempting to read a meniscus on a quart or liter size container.

Because formaldehyde is prone to oxidation, it should be mixed with distilled or deionized water. Tap water usually contains unwanted chemicals that will affect the quality of the mixed solution. However, in the field it is often necessary to use local water sources, which is why it is important to record in your field notes the source of formaldehyde and water used.

Formaldehyde Safety

Safety precautions must be observed when working with formaldehyde both in the field and the laboratory (Simmons 2014). Most people who work with formaldehyde for any length of time develop sensitivity to it. Formaldehyde is an irritant to the skin, mucus membranes, and the respiratory system, and presents a cancer risk with improper exposure (see review in Simmons 2014). According to established safety standards, if you can smell formaldehyde, you are receiving an unsafe dose (OSHA 1992).

- Use formaldehyde only in a well-ventilated area.
- Always wear eye protection (splash goggles that meet ANSI Z87.1 1989 standards) while working with formaldehyde, as it is easily absorbed by mucous membranes, and may cause serious damage if splashed in the eye.

- Do not wear soft contact lenses when working with formaldehyde. Soft lenses can absorb formaldehyde and trap it against the eye.
- Wear gloves made from neoprene or other material rated for formaldehyde resistance. See *Appendix III* for sources of safety equipment, including rated resistant gloves and eye protectors. Do not use latex gloves—formaldehyde readily penetrates latex (Raloff 1985).

Bear in mind that most people are regularly exposed to formaldehyde from a wide variety of common products ranging from cosmetics to cigarettes to insulation to plastics. This environmental formaldehyde exposure in addition to laboratory or fieldwork exposure means that even a short-term exposure in the field or laboratory can lead to a dangerous accumulative exposure.

Paraformaldehyde

The solid polymer of formaldehyde, called paraformaldehyde, may be carried in the field as a dry powder, flakes, or pellets, and mixed with water and a buffer for use. Taub (1962) suggested mixing paraformaldehyde with sodium hydroxide in the field, but hot to boiling water is needed to dissolve the paraformaldehyde, and this method results in a formaldehyde solution with a pH of 12. Huheey (1963) suggested replacing the sodium hydroxide with sodium carbonate, which eliminates the need to boil the solution, but produces formaldehyde with a pH of 11.6, which is still so alkaline that it will likely cause specimens to clear. Nelson and Sparks (1999) cautioned that if Alconox is used to dissolve paraformaldehyde, the solution may cause specimens to clear, lose pigment, and swell. These problems might be avoided by a thorough rinsing between fixation and preservation in alcohol.

Ehmann (1989) suggested a more practical method for field preparation of paraformaldehyde, by preparing the ingredients in advance in two plastic bags. In one plastic bag, mix 80g of paraformaldehyde (flakes or powder), 20 g of anhydrous sodium carbonate, and 0.5 g of a wetting agent in solid form (e.g., a detergent). In a second plastic bag place 21 g of citric acid. In the field, mix the contents of the first bag with 2 liters of water, stirring constantly to prevent clumping, and agitate the solution occasionally over the next four hours at 20–30°C to dissolve the contents. Once the paraformaldehyde has dissolved, add the citric acid and stir until no carbon dioxide evolves (about five minutes), then set the solution aside and allow gas to continue to escape from it over the next 24 hours. The resulting mixture will be 10% formalin that is neutrally buffered and ready for use. However, it is possible that some wetting agents may contain chemical contaminants that could cause problems in later use of the specimens.

PRESERVATION

Good preservation depends on the rapid penetration of a fixative (usually formaldehyde) or preservative (usually ethyl alcohol) into the tissues. Four basic methods are used to introduce the fixative or preservative solution into tissues.

1. ***Opening the Body Cavity.*** The oldest method is to make cuts into the body cavity and into large muscle masses, particularly on the limbs, to allow the chemicals to penetrate. This method is not desirable because of the damage it does to the specimen, but may be used in an emergency when no other option is possible.

2. **Perfusion.** The most efficient method of distributing the preserving chemicals to the tissues is by perfusion (Jones and Owen 1987; Quay 1974; Schultz 1924). A cut is made for access to the left femoral artery or the left ventricle of the heart for injection of the fixative or preservative; a second cut is made to open a vein to provide an exit for the fluid. Insert a large (e.g., 18–24 gauge) needle into the artery, hold it firmly in place, and apply gentle pressure. Force fluid through the system until the effluent is clear, or without blood. A large syringe (20–50 cc capacity) holds sufficient fluid to perfuse most small amphibians or reptiles without refilling. For larger animals, a valve may be used to permit refilling of the syringe without allowing air to enter the system, or a pump may be used to draw solution from a large container. The drawbacks to this method include that two cuts must be made in the animal, the blood is replaced by the preserving fluid, and the process takes a little more time to complete than traditional preservation.
3. **Permeation.** Smaller specimens may be placed directly on formaldehyde-soaked paper towels for hardening, then covered with more paper towels and a volume of formaldehyde (see *Hardening and Tagging*). Sufficient formaldehyde should penetrate the skin of fresh specimens rapidly. Larger and thicker-skinned specimens must be injected (see *Injection*). As a general rule, any amphibian smaller than about three inches long (snout-vent length) should not have to be injected for the formaldehyde to penetrate the body tissues, except for gravid females. Most reptiles must be injected, due to their thicker skin. Of the fixatives currently available, only formaldehyde has sufficient power of penetration to be used with whole animals. Most histological fixatives, including glutaraldehyde, are inadequate for permeation of whole animal bodies (see review in Simmons 2014).
4. **Injection.** Injecting the fixative solution into the body cavity and the major muscle masses is the standard procedure now in use for nearly all reptiles and for larger amphibians. Do not inject too much formaldehyde in any one part of the body. The ability of formaldehyde to penetrate tissue is quite good, once it is through the skin. More rapid injection of fixative may be attained by attaching a valve and supply tube to the syringe, by using a pump action spray bottle or hand pump (Congdon et al. 1975; Jackson 1971; Johnson and Parker 1980; Saul 1982), a small electric pump, or a garden sprayer (Forstner et al. 1997). Inject as detailed below. Take care to avoid being splashed by formaldehyde should the needle or supply tube come loose or a syringe plunger leak.

Large Amphibians. Inject formaldehyde into the limbs and body cavity. Amphibian eggs and larvae do not need to be injected, but should be submerged in the fixative or preservative.

Reptiles. One of the pair of hemipenes should be everted in males before injecting or perfusing the body. Using a syringe of fixative or preservative, insert the needle on the left side of the base of the tail, and put your thumb over the right side of the tail and vent to prevent both hemipenes from everting. Apply gentle pressure until the hemipenis is everted, then tie a soft string or thread around the base of the hemipenis so that it remains extended.

Lizards should be injected in each limb, in the body cavity, and anteriorly into the gular region. Using a very sharp needle, make a series of perforations through the skin of the tail from base to tip on the ventral surface to allow the solution to penetrate, or make a series of perforations on the dorsal and lateral surfaces of the tail (L. Vitt, pers. comm.). Geckos and other specimens that are coated with a thin, waxy film may pose problems for the penetration of fixatives, and the separation of the stratum corneum from the lower epidermal layers. Such specimens may first be soaked for a couple of minutes in a 70% ethyl alcohol solution to remove the waxy outer coating, then placed in the hardening tray (L. Grismer, pers. comm.), or a small series of perforations with a sharp needle may be made on the limbs, body, and tail (L. Vitt, pers. comm.).

Snakes should be injected ventrally at several points along the length of the body cavity. Very large specimens should be injected at several points along either side of the spine down the full length of the body and tail. Make a series of perforations through the skin of the tail from base to tip on the ventral surface to allow the solution to penetrate.

Turtles. Extend the head and neck from the shell. Insert a small piece of soft wood, plastic, or cork in the mouth to keep the jaws open. Inject the preservative into the neck, limbs, tail, and deep into the body cavity.

Reptile Eggs. Reptile eggs may be submerged in 10% buffered formaldehyde or 70% ethyl alcohol for preservation. If the embryos appear to be near term, they may be injected through the eggshell using a thin needle, or small slits may be cut in the eggshell.

Preparing the Hardening Tray

Specimens should be laid out in a flat-bottomed tray for hardening. Use fairly shallow trays with well-fitting lids. I prefer trays made of polypropylene with snap-top lids. These are relatively unbreakable, resistant to fixatives and preservatives, and keep the formaldehyde fumes contained. Do not use metal trays, because the formaldehyde will oxidize the metal and cause the specimens to discolor. Line the bottom of the tray with a layer of paper towels and moisten them with buffered 10% formalin solution. Smooth out the paper towels to remove the air bubbles and creases, providing a flat surface on which to position the specimens. Do not use paper towels moistened only with water for this step. Although this is a way to avoid contact with formaldehyde vapors, there are two serious drawbacks—the specimens may jerk and twist into contorted positions as the formalin solution is added, and the paper towels moistened with water will dilute the formalin.

In an emergency, where no hardening tray is available, specimens can be fixed and hardened in plastic bags (Hahn 1959). A hardening tray for very large specimens can be fashioned from a large sheet of plastic, using boards or bricks to hold up the sides, by lining a pit in the ground with a plastic sheet (Darovec 1979), or by lining a cooler with plastic (L. Grismer, pers. comm.).

Positioning Specimens for Hardening.

Specimens are positioned for hardening in a natural posture that allows (1) key features of the specimen to be accessed readily; (2) the maximum

number of measurements to be obtained easily from the specimen; (3) the specimen to be inserted and removed from a container without tangling up with other specimens; and (4) packed for shipping without damage to digits or tails (Fig. 13).

Metamorphosed amphibians are positioned in the hardening tray on their venter, with limbs positioned in a natural relaxed manner and fingers and toes spread to show webbing (Fig. 13). Large limbless amphibians (e.g., caecilians) may be positioned as are snakes (see *Snakes*); long amphibians (such as certain salamanders) may be positioned as are lizards (see *Lizards*).

Amphibian larvae and eggs should be placed in a vial (Fig. 14) or flat-bottomed jar with the buffered 10% formalin solution and the field tag (Bragg 1949). Lay the container on its side if necessary so that the specimen will harden in a flat position. Do not overcrowd specimens. When eggs are attached to vegetation, include a small piece of the vegetation with the eggs as well. Removing the adherent vegetation will damage the eggs, and it is important to know whether the clutch is affixed to a stem or leaf.

Lizards are placed on their venter with limbs flexed in a natural position and toes slightly spread (Fig. 13). If present, the dewlap (Fig. 15) may be extended to one side and held in place with the lizard's hand while hardening. The caudal appendage of long-tailed individuals may be curled up around the body.

Snakes. Small snakes are coiled clockwise with the head to the outside in a coil that will fit a standard size jar. Snakes too large to coil in the bottom of a standard size jar should be coiled up the inside of a container the same size as a standard storage jar, with the head on top.

Turtles should have the limbs, head and neck, and tail extended from the shell. The mouth should be held open during hardening with a piece of soft wood, plastic, or cork. It is sometimes necessary to pin the limbs and neck in position until the specimen is hardened.

Very large reptile specimens are often not preserved whole due to the expense of preserving fluids and transportation. If this is the case, the body should be carefully measured, and the skin removed, preferably with the head and tail entire. The remaining body should be roughed out (gutted and major muscle masses removed—see *Preparing Specimens for Skeletonizing*) and dried for later preparation as a skeleton. Take photographs and careful notes on stomach contents, guts, reproductive state, and size of fat bodies before disposing of the remains. For hardening, lay the skin out flat, dorsal side down. Cover the entire inside surface of the skin (including inside the limbs) with a layer of cheesecloth, which will serve as a wick to draw the fixative or preservative in. Roll the skin up loosely and submerge it in the fixative or preservative solution. An alternative is to disarticulate the specimen and preserve it in pieces (Cook 1965).

Hardening and Tagging

Once positioned in the hardening tray, specimens are covered with a layer of paper towels dampened with fixative, and a standing amount of buffered 10% formalin solution is carefully poured over them. Put the tray in a place where



Figure 13. Positioning specimens for hardening.

it will not be moved for several hours. Small amphibians will harden in a few hours; larger specimens and most reptiles will require overnight.

Field tags may be affixed to the specimens before they are positioned for hardening, or after they have hardened. Tie tags on the specimens with a soft cotton thread. On animals with limbs, tie the field tag on the animal's left hind leg. If the left hind leg is not present, or is damaged, tie the tag on the animal's right hind leg, or around its waist. If the animal is so small that tying on the tag might damage the leg, tie it around the animal's waist. For very tiny animals, place the tag and the animal together in a small vial without tying on the tag. Tags should be tied tight enough that they will not come off when the animal is handled, but not so tight as to cause distortion to the body. For snakes, caecilians, and other limbless animals, sew the tag through the neck, approximately one head length behind the constriction of the neck, but take care to sew below the vertebral column so the vertebrae are not damaged.

For eggs or larvae, place the tag in the vial with the specimens so that the field number may be read from the outside. Wrap tape around the vial/lid junction to keep the lid from becoming loose during transport.



Figure 14. Tadpoles in vial, with David McLeod (Sakaerat Environmental Station, Thailand).



Figure 15. Dewlap of *Anolis aequatorialis* (3 km SW Tandayapa, Pichincha, Ecuador, 1759 m).

After the specimens have hardened and the tags are affixed, transfer the specimens to a large container where they can reside submerged in the 10% buffered formalin solution for a few days before packing or further processing. Leaving specimens in formalin solutions for too short a time will result in incomplete preservation; leaving specimens in formaldehyde longer than a few days will cause darkening and loss of pattern in specimens as well as pose a risk of decalcification of bone (Simmons 2014; Stuart 1995).

PACKING AND TRANSPORT OF SPECIMENS

Sad experience taught us but too late, that from the sultry humidity of the climate, and the frequent falls of the beasts of burden, we could preserve neither the skins of animals hastily prepared, nor the fishes and reptiles placed in phials filled with alcohol.

—Alexander von Humboldt, 1889

After specimens have been in formaldehyde for a few days, they may be packed for shipment. Lay specimens out on a length of cheesecloth as described for packing specimens for loans (see *Procedures for the Preparation Outgoing Loans*). Keep the size of the package per plastic bag small—this will make packing easier, and minimize loss should leakage occur. Do not leave standing liquid in the plastic bags. Double bag each package, closing the tops of the bags securely. Include an address label between the outer bag and inner bag of each package. Label each bag as “Preserved scientific specimens—No commercial value.”

Larvae, particularly small larvae, should be transported in liquid. Transfer the specimens to the smallest size vial that will hold them without causing distortion. Diluted formalin or water may be used if shipping by air, but never put larvae in alcohol. Fill the vial completely with the liquid and secure the lid tightly to prevent leakage. Pad the vials with paper towels, and enclose them in a plastic bag. Label each bag as “Preserved scientific specimens—No commercial value.” Upon arrival, transfer the larvae to larger vials and a fresh buffered 10% formalin solution.

Ship specimens by air whenever possible to save time (see *Shipping Fluid-preserved Specimens* for details). Upon arrival, specimens should be unpacked and prepared for permanent storage (see *Part IV, Museum Collections*).

PREPARING SPECIMENS FOR SKELETONIZING

Reptile specimens to be prepared as skeletons by the use of arthropods (e.g., dermestid beetles) should be roughed out (gutted and most muscle mass removed) and the carcasses dried rather than formaldehyde fixed. One way to easily desiccate a carcass is to hang it in a wire basket in the engine compartment of a field vehicle (Bond 1939) or dehydrate it in ethyl or isopropyl alcohol (L. Grismer, pers. comm.). Amphibian specimens should be preserved but not desiccated before skeletal preparation as desiccation will cause warping and cracking of cartilage (Simmons 1986a). Skeletons should be prepared from properly identified individuals with good locality data. Record the animal's sex, snout-vent length, breeding condition, and any other pertinent information in your field notes.

EMERGENCY SUBSTITUTES FOR FIXATIVES AND PRESERVATIVES

Sometimes an opportunity arises to preserve specimens when formaldehyde is not available or its use is not desirable. In this case, 70% ethyl alcohol is the best substitute. Do not use full-strength alcohol to preserve specimens, as this will cause excessive dehydration of the tissues. If ethyl alcohol is not available, then 40–50% isopropyl alcohol may be used (see discussion of alcohols in *Selection of a Preservative*). Methyl alcohol does not make a satisfactory preservative (Simmons 2014). If neither ethanol nor isopropanol is an option, use the strongest beverage alcohol available. I have been given specimens in Ecuador and in Paraguay that were preserved with locally available aguardiente (a beverage alcohol made from sugarcane and usually flavored with anise; it typically has an alcohol content of about 40%). In both cases, the specimens had a rather distinctive odor years later, but they certainly qualified as preserved.

COLOR PRESERVATION

Both reptiles and amphibians will fade after fixation in formaldehyde or preservation in alcohol. Several formulas to better retain colors have been proposed, but none has been proven effective in the long term, either for color preservation or specimen preservation, and their use is not recommended. For recipes see Guerra (1976); Waller and Eshmeyer (1965); White and Peters (1969); and Windsor (1971). Colors can also be preserved to some extent by preserving the skin as a dry preparation (see Beebe 1947; Juszcyk 1952; Kincaid 1948; Paranjape and Mulhekar 1979; Schueler 1981; and Taylor 1981b).

FROZEN SPECIMENS

Once a specimen has been frozen, it is usually not possible to make a satisfactory fluid preserved preparation from it (Scott and Aquino-Shuster 1989; Simmons 2014). Freezing causes cells to rupture, weakening the tissue (Mills 1975). Fixatives and preservatives cannot penetrate tissues while they are frozen, and tissues will begin to break down as they thaw. Frozen specimens are best prepared as skeletons. If it is necessary to make a fluid preparation from a frozen specimen, thaw it in a solution of buffered 10% formalin, injecting it as thawing permits.

MISCELLANEOUS PRESERVATION TECHNIQUES

General Preservation Techniques for Amphibians and Reptiles.

Anderson (1965); Casas-Andreu et al. (1991); Knudson (1972); Scrocchi and Kretzschmar (1996).

Specific Types of Preparations. Embedding in plastic (Robbins 1976); embalming (Elkan 1970); freeze drying larvae (Altig (1975); hemipene preparation (Dowling and Savage 1960; Klauber 1997; Ortenberger 1923); paraffin infiltration (McCoy 1929; Noble and Jaeckle 1926).

Skeletal Preparation. Amphibian skeletons (Simmons 1986a—do not use bleach as recommended in this paper); clothes moth larvae (Banta 1961); dermestids (Borrell 1938; Jannett and Davies 1993; Sommer and Anderson 1974; Tiemeier 1940; Valcarcel and Johnson 1981; Vorhies 1948); dermestids

with specimens previously preserved in fluid (De la Torre 1951; Dirrigl et al. 1993; Gritis and Brunner 1990); how to skin a giant tortoise (Anon. 1899); maceration (Fenton et al. 2003; Hill 1975); mealworms (Allen and Neill 1950); preparation of turtle shells (MacMahon 1961); terrestrial isopods (Maiorana and Van Valen 1985).

Clearing and Staining. Campbell (1986); Davis and Gore (1936); Dingerkus and Uhler (1977); Maisano (2008); Potthoff (1984); Springer and Johnson (2000); Taylor (1967a, 1967b); Taylor and Van Dyke (1985); Wassersug (1976).

PART IV. MUSEUM COLLECTIONS

A major systematic collection of amphibians and reptiles is like a three-way hybrid between a pickle warehouse, a reference library, and a mail-order establishment.

—C. J. McCoy (1981)

Collections not used

Are useless collections

—I. P. Sofacto (by way of Robert Waller)

The most important functions of collection management are (1) maintaining the integrity of the specimens and their associated data; (2) maintaining the specimens and data in optimum usable condition; and (3) making the specimens and data available for appropriate use.

Collections are useless if the specimens sit out the passing decades untouched on dusty shelves. To be valuable resources, specimens must be made available to the scientific community and other appropriate users, while at the same time the integrity of the collection is maintained. Data files should be cross-referenced and kept up to date with the addition of specimens and refinement of specimen-related information; the storage environment and the physical condition of the individual specimens must be monitored regularly; collection use must be evaluated and documented.

In order to maintain the integrity of the specimens and data, standardized operating procedures (SOPs) should be established, documented, and implemented for the various tasks undertaken in the course of management. The use of standardized procedures will insure that different individuals working with a collection will be able to maintain its continuity. Write down the standardized procedures you put into place in the collection, and keep a written record of any changes in these procedures. How you do things today may well be important to someone using the collection in 50 or 100 or 200 years. Because you can never predict with certainty when you will be able to resume an interrupted procedure, it is important that it be obvious to another worker what you were doing and how you were doing it.

Despite the fact that herpetological collections have continued to grow due both to field collecting and because many institutions have taken on orphaned collections, there has been no significant corresponding increase in the number of collections care workers to manage these resources. Nevertheless, Simmons and Muñoz (2003) have demonstrated that as a collection increases in size arithmetically, the costs of managing the collection (particularly the amount of time that must be invested in collections care) increases geometrically.

CONSERVATION OF HERPETOLOGICAL COLLECTIONS

Historically, the conservation of specimens and data-bearing media has received very little attention in natural history collections. Fortunately, over the last 20 years, much information about materials and techniques (e.g., Rose and Hawks 1995; Williams 1999), standards for collection care (Simmons 2013, 2015; Society for the Preservation of Natural History Collections 1994),

laboratory safety (Hawks et al. 2010), and fluid preservation (Simmons 2014) has become available.

Conservation means the preservation of a specimen in a way that retains as completely as possible its original composition while prolonging its useful life as a specimen. Conservation concerns for herpetological collections begin with the chemicals and techniques used to capture, euthanize, and preserve the specimen, and continue with the collection storage environment and how the specimens are used.

Natural history specimen preservation and collection management is plagued by a long tradition of untested techniques, handed down from one worker to another, and numerous anecdotal observations that seem to explain phenomena but are often baseless. It is important to distinguish between those processes and techniques that have been appropriately tested or are known to contribute to the long-term stability of the specimens and those which are part of a dubious oral tradition. Many techniques reported in the literature appear to work because the damage they do takes decades to become apparent. For example, consider phenoxetol, enzyme detergents, and formaldehyde. Phenoxetol was used with apparent success for ten years before its failure as a preservative was determined (Crimmen 1989). It took 20 years before the use of enzyme detergents to prepare skeletons was shown to be a destructive practice (Shelton and Buckley 1990). The use of formaldehyde fixation has never been tested against the use of alcohol preservation without formaldehyde (Simmons 2014). Many traditional collections care practices (such as replacing preservative solutions when they become slightly discolored, or mixing preservatives with tap water) negatively affect the useful life of the specimens.

Fluid-preserved Specimens

The long-term stability of a fluid-preserved specimen depends chiefly on three factors: (1) the quality of initial fixation and preservation; (2) post-preservation processing; and (3) the quality and stability of the storage environment (Table 2).

The biggest threats to fluid-preserved herpetological specimens are inadequate preservation, loss of fluid preservative, and exposure to heat, light, and ultraviolet radiation (Table 2). Unfortunately, the choice of fixatives and preservatives relies mostly on tradition, not empirical knowledge (Simmons 1993, 1995). Most herpetologists believe that formaldehyde fixation is necessary for the long-term preservation of specimens, but formaldehyde has only been in use since the 1890s, while the oldest known fluid-preserved specimens are more than two hundred years old. While formaldehyde fixation is necessary to preserve some types of specimens and for many histological preparations, no published controlled experiments demonstrate that formaldehyde fixation enhances the long-term preservation of most specimens.

Skeletal Specimens

Most skeletal preparations are either dry or in the form of cleared and stained specimens stored in glycerin. References to techniques for preparing

Table 2. Factors determining the long-term usefulness of fluid preserved specimens.

1. Quality of the initial fixation or preservation	a. Time interval between death and treatment (fixation or preservation)—should be as short as possible.	
	b. The quality of the fixative or preservative—mix and measure carefully, using deionized or distilled water.	
	c. Rate of penetration of the fixative or preservative into the tissues.	
	d. Temperature of fixation or preservation (around 35°C may be the best temperature for fixation).	
	e. Proportion of fluid preservative to specimen volume (should be at least 7:3).	
2. Post-fixation Processing	a. Washing time—keep as short as possible.	
	b. Transfer to preservative—use steps of 20% for transfer to preservatives ≤ 70 .	
	c. Hydration or dehydration of specimen.	
3. Storage Environment	a. Temperature and fluctuations—18–21°C is preferred, minimize fluctuations.	
	b. Light (intensity and cumulative exposure)—reduce as much as possible.	
	c. Ultraviolet radiation (intensity and cumulative exposure)—reduce as much as possible.	
	d. Relative humidity and fluctuations—ca. 50% is preferred, minimize fluctuations.	
	e. Vibrations—minimize.	
	f. Integrity of the container seal.	i. Preservative evaporation.
		ii. Contamination of the preservative.
	g. Handling and use of the specimen.	i. Physical damage.
		ii. Dehydration.
		iii. Rehydration.

skeletons are listed in *Miscellaneous Preservation Techniques*. Dry skeletons are susceptible to attack from pests, so they must be monitored regularly, as part of a program of integrated pest management (IPM). Using IPM techniques, most pest problems can be prevented or dealt with without the use of chemicals (Jessup 1995; see also *Appendix III*).

Skeletons should not be boiled, bleached, subjected to ammonia or solvent treatments, or treated with consolidants or whiteners. Studies have shown

that these treatments may cause premature aging of bone, interfere with chemical analysis such as DNA extraction, or cause later deterioration of bone (Caldararo and Grabow 2000; Shelton and Buckley 1990; Williams 1999). Sometimes bones become greasy or chalky in storage. Chalkiness results from prior treatment with enzymes, bleach, ammonia, or other chemicals that were not adequately rinsed out of the bone. There are no treatments to correct this deterioration. The grease that migrates to the surface of bone is a natural component of the bone. Some animals, such as sea turtles, have particularly greasy bones. Removal of grease from bones is done for cosmetic purposes, not to make them better scientific specimens. Degreasing is an unnecessary treatment that damages bone. Typical degreasing treatments include boiling the bones, subjecting them to ammonia baths, and boring holes for the penetration of solvents, all of which damage the bone and reduce its scientific usefulness. Grease accumulation on the surface of bones may make some features difficult to see, may allow acidic or abrasive particles to adhere and damage the bone, and may attract pests. If grease poses a risk, clean the surface of the bone with 95% ethyl alcohol using polyester fiber or cotton swabs. If the grease continues to accumulate, another surface cleaning can be done later. When housing skeletons with a high grease content (e.g., marine turtles) take measures to prevent the grease from damaging tags, labels, the substrate the bones are sitting on, and adjacent specimens (e.g., enclose the greasy skeletons in polyethylene bags).

The concerns for glycerin preparations are similar to those for other fluid-preserved specimens, with a couple of added precautions. Glycerin is hygroscopic, so it absorbs moisture from the air. If the containers are not tightly closed, when relative humidity is high the glycerin may absorb sufficient water to overflow the container, and may even perilously dilute the preservative solution. Along with moisture from the air, glycerin absorbs atmospheric pollutants and contaminants that may cause it to become too acidic or alkaline to serve as a good preservative. In addition, glycerin is an excellent growth medium for bacteria and mold. Containers of glycerin preparations must be well sealed, and a small amount of thymol ($C_{10}H_{14}O$) or camphor should be added to each container to prevent bacteria and mold growth (see *Procedures for Handling Specimens in Glycerin*).

Genetic Resources

Samples for genetic analysis are generally frozen and stored in liquid nitrogen or ultra-cold freezers. Refer to Gamble (2014) for guidance on the collection, preparation, storage, and curation of genetic material, and to Zimkus and Ford (2014) for best practices for the management of genetic resources.

The Storage Environment for Herpetological Collections

The single most important factor for the longevity of natural history collections is the quality of their storage environment. This means maintaining the most stable temperature and relative humidity possible (with minimal fluctuations from seasonal set points), and protecting the collection from light and ultraviolet radiation. In general, a temperature of 65–70°F (18–21°C) and relative humidity of around 50% is preferred for the storage of natural history collections (Appelbaum 1991; Bachmann and Rushfield 1992). The optimum temperature for storing fluid-preserved specimens—cool

enough to reduce the flash point and evaporation of the preservative without affecting preservative or specimen quality—is 65°F (18°C) at 50% relative humidity (Simmons 2014). Higher temperatures increase the rates of the chemical processes of deterioration (Rose and Hawks 1995; Thompson 1986); lower temperatures may cause problems with preservative quality (Simmons 2014). Freezers and refrigerators should not be located in the same rooms as fluid-preserved collections, both because electrical equipment increases the risk of fire, and because compressors generate heat, making it more difficult to maintain the collections at the correct, steady temperature.

All materials in the collection are affected by fluctuations in temperature and relative humidity—the paper that the field notes, labels, and catalogs are written on expands and contracts, breaking along binding seams; skeletal material is stressed; teeth loosen and fall out of skulls; jar lids become loose; the emulsion peels from photographs; inks fade (Browning 1970; Shahani and Wilson 1987; Thompson 1986). Relative humidity and temperature should be monitored in the collection area throughout the year with electronic data loggers (see *Appendix III*); based on this data, steps should be taken to ameliorate the environmental fluctuations that occur in collection storage.

Preserved specimens should be kept in the dark whenever they are not in use. Lights in the collection storage area should be turned off when not in use. Fluorescent lighting in particular produces significant amounts of ultraviolet (UV) radiation, which is very damaging to specimens. All UV sources in specimen storage or use areas, including laboratories and offices, should be equipped with UV filters (see *Appendix III*) or eliminated. Glass jars do not provide adequate protection from UV—glass allows damaging radiation in the 310–400 nm range to pass through (Harris 1968; Macleod 1975). Preserved specimens should never be exposed to sunlight or ambient light coming in through a window.

Specimens in fluid should be kept in glass or stainless steel containers with good closures. Because alcohol-preserved specimens are hygroscopic, when specimens are removed from their containers to be examined, they should be kept submerged in a tray of the same preservative they are stored in. Do not put alcohol-preserved specimens in water for examination because water absorption causes extreme stress to the specimen, and the absorbed water will dilute the preservative when the specimen is returned to its container. The cycle of water absorption and subsequent dehydration in the preservative will cause permanent damage to preserved specimens and dilution of the preservative in the storage container. Formaldehyde-preserved specimens may be submerged in water for examination—because formaldehyde-based preservatives are mostly water, the water submersion will not damage specimens or significantly dilute the preservative.

Skeletons, turtle shells, microscope slides, magnetic tapes, photographic negatives and prints, color transparencies, compact disks, and other digital storage media should be protected from dust and environmental fluctuations in closed containers made of acid free board, polyethylene, polypropylene, or polystyrene, or in metal cabinets that have a powder coat finish. Many dust particles are acidic or abrasive (Thompson 1986). Combined with moisture

from the air or with smoke (which is oily), dust can form a coating that will etch the surfaces to which it adheres.

Because heat greatly accelerates the deterioration of cellulose (Appelbaum 1991; Banks 1978; Browning 1970), the cooler the storage temperature the better for paper, above a lower limit of 60°F (15°C). Paper-based collection records should be housed in a stable storage environment and protected from light, ultraviolet radiation, and pollutants. The ideal storage environment for most paper is 68°F (20°C) $\pm$ 4°F (2°C) at a relative humidity of 45% $\pm$ 2% (Shelley 1992; Vogt-O'Connor and van der Reyden 1996). Refer to Vogt-O'Connor and van der Reyden (1996) for a summary of information on the storage of paper-based materials.

Important papers that are deteriorating (e.g., documents on acidic paper, mimeographed documents, self-carbonized forms) should be scanned to preserve an electronic copy, or copied using preservation photocopy techniques. Refer to Jordan (1993) for recommendations on equipment, materials, and procedures for preservation photocopying.

Field notes and other permanent collection documentation should be protected in acid-free slipcases or enclosures (Thompson 1986). Refer to Vogt-O'Connor and van der Reyden (1996) for a detailed discussion of the storage of paper-based materials. Binding field notes with acid-free materials is acceptable only if existing holes in the pages are used. Binding methods such as the standard "library binding" (side-seam sewing) are not acceptable (Banks 1978) because the paper develops a stress line where it is sewn, and with use, will break along this line. When bound field notes are opened to be read, the pages are pulled taut against the threads, cutting and tearing the paper. When the bound notes are forced down on a flat photocopier glass, the damage is worse. Library bindings are designed to protect materials from a lot of handling over a short period of time, not for long-term archival protection of permanent documents.

Photographs, negatives, and color transparencies should be stored at 68°F (20°C) $\pm$ 4°F (2°C) at a relative humidity of 20-45% for prints and 20-30% for negatives, with fluctuations of no more than $\pm$ 3% (Henricks 1992a; Vogt-O'Connor 1997). Wear cotton gloves when handling photographs and negatives. Albright (1993) provides recommendations for proper enclosures for prints and negatives. Color transparencies deteriorate much faster than prints or negatives—the only way to prolong their useful life for more than 10-20 years is to make copies for use and keep the originals in cold storage. Refer to Vogt-O'Connor (1997) and Wilhelm and Browner (1993) for more information on caring for photographic materials. Store photographic materials away from light.

Recorded magnetic tapes must be stored so that the tape is not distorted. Avoid winding the tape too tightly by storing the reel after it comes off the machine at playback speed (Hendricks 1992b, Wickstrom 1982). Store tapes and other magnetic media in an area protected from airborne particulates and extremes of temperature or rapid changes in relative humidity. The ideal storage environment for magnetic media (e.g., tapes, computer

disks, external hard drives, flash drives) is 49–59°F (9–15°C) at a relative humidity of 25–45%, with fluctuations of no more than 3% (Riss 1993; Vogt-O'Connor 1996). The useful lifespan of magnetic media is about 10–30 years. Originals of magnetic media should be maintained in cold storage with use copies made available for regular access (Riss 1993; Vogt-O'Connor 1996). Spools of tape should be rewound once every six months to a year at playback speed to reduce print-through and the uneven tension in the roll that builds up from ordinary environmental fluctuations. Refer to Henricks (1992b); McWilliams (1979); Riss (1993); van Bogart (1995); and Vogt-O'Connor (1996) for further discussion of tape and equipment for its storage and protection.

Recorded tapes and computer disks should be kept in archival quality boxes and stored vertically, not horizontally or at an angle, to minimize data migration and bleed-through. Protect these objects from heat, light, magnetic fields (including electric motors and audio speakers), and air-borne contaminants. Playback heads on tape equipment should be cleaned regularly and demagnetized after every 20 hours of use.

Monitoring the Storage Environment

Environmental monitoring should be done in all collection storage areas. Dataloggers are more cost-effective than hygrothermographs, respond faster to changes in the environment, and provide data in electronic format that is easier to analyze than the paper charts produced by hygrothermographs. If recording equipment is not available, an inexpensive thermometer and humidity indicator can be purchased, read two or three times each day, and the data recorded manually. Environmental records should be archived as part of the permanent documentation of the collection. These records are important for evaluating storage conditions, quality of preservation, and rates of deterioration.

THE COLLECTION STORAGE FACILITY

The collections care staff should have complete control over access to collection storage areas. To increase environmental stability, collection storage areas should not have windows, outside walls, and should not contain offices or work stations. The shelving and floor loading must be adequate to accommodate the weight of the containers (a one-gallon jar of alcohol with specimens may weight more than eight pounds). Moveable shelving (compactors) may be used to maximize space (Fenner 1992). If shelving is constructed more than five shelves high, a safety ladder will be needed for access. Switches should be wired so that lights can be turned on selectively in each aisle so that when one area is being utilized, it is not necessary to illuminate the entire room. Ultraviolet filters should be installed on all UV producing light sources. Suspended light fixtures must be high enough to not interfere with access of containers on the shelving. Shelves should be equipped with lips and restraining bars (earthquake bars). Colbert (1961) described a sturdy shelving system using wooden shelves on steel frames. Metal shelves should be made of stainless steel, as no coatings applied to metals (e.g., epoxy, paint, electrostatic powder coating) are sufficiently resistant to the friction caused by movement of specimen containers.

Safe Storage and Handling of Fluid-Preserved Specimens

The federal government closely regulates the acquisition, storage, and use of ethyl alcohol. A permit must be obtained from the Alcohol and Tobacco Tax and Trade Bureau National Revenue Center to purchase and dispense tax-free ethyl alcohol (see *Appendix III*). Strict storage and use guidelines for alcohol must be followed. Isopropyl alcohol may be purchased and dispensed without a federal permit.

Denatured Alcohol and Alcohol Additives

Ethyl alcohol that has been denatured may be purchased without a permit. Alcohol is denatured by the addition of chemicals to make it unsuitable for human consumption. Common denaturants include acetone, methanol, and various ketones (Simmons 2014). It is often difficult to determine the denaturants or their concentration, and suppliers may change denaturants without notice. All denaturants have the potential to render specimens unfit for future chemical analysis, thus denatured alcohol is considered undesirable as a preservative. When working with old alcohol-preserved specimens, keep in mind that other substances may have been added to alcohol preservatives in the past in an attempt to enhance the utility of the preservative. The most common additives are glycerin, acetic acid, and formaldehyde, but such compounds as alum, arsenic, and mercuric chloride may also be encountered (Simmons 2014, Simmons et al. 2007). Alcohol preservatives with these or other additives should not be used for specimen preservation.

Safety

Two main safety issues are of concern in fluid-preserved collections. The first is to ensure that the collection is stored and handled in ways that are safe both for the specimens and for the collection care workers. The second consideration is compliance with fire codes and hazardous chemical regulations.

Safe storage of preserving fluids begins with using containers that provide adequate seals, and keeping the containers on appropriate shelving. The collection storage area must be adequately ventilated. Alcohol and formaldehyde are absorbed through the skin and the mucous membranes of the body, particularly through the lungs (see *Formaldehyde Safety*). Work areas should have bench-top fume collectors positioned so that the fumes are drawn directly from the mouths of containers or from trays of specimens (Fig. 16). Open containers of fluid-preserved specimens only in areas with proper ventilation.

A very small amount of evaporation of fluid preservatives may occur in a fluid collection, either by gas escaping from containers due to changes in temperature and air pressure, or when containers are opened to access specimens. Work areas and storage areas must have sufficient airflow to prevent the accumulation of preservative fumes. Because alcohol is heavier than air, alcohol fumes accumulate at floor level, so the ventilation system must function at floor level. Keep equipment that produces sparks, flames, or heat away from collection storage areas. A strict no-smoking policy must be enforced in the collection storage and work areas. Class A-B-C fire extinguishers should be available in both work and storage areas.

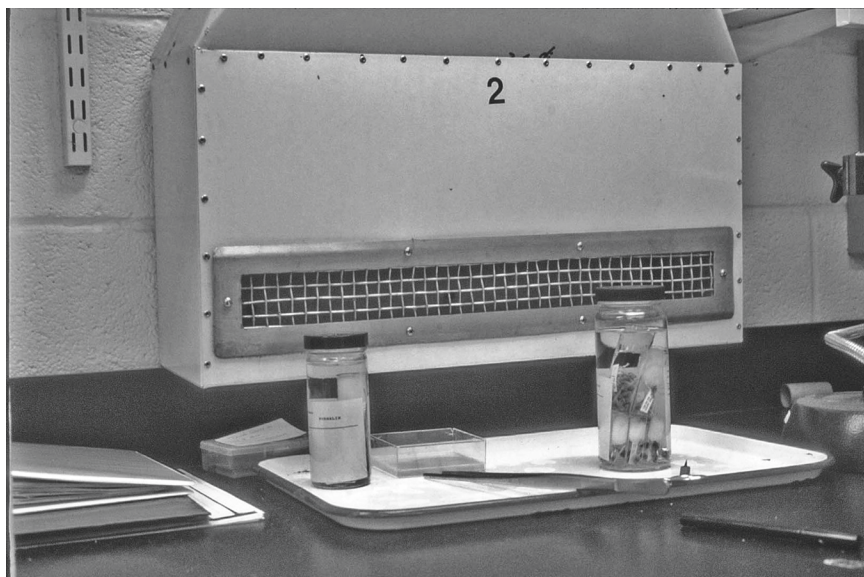


Figure 16. Benchtop fume collector.

Personnel working with fluid-preserved specimens should wear lab coats, neoprene gloves, and either safety glasses or splash goggles as appropriate to the task. Open-toe shoes should not be worn in laboratory areas.

Fire Code and Safety Regulations

When the vapors from flammable or combustible liquids mix with air and come into contact with an ignition source, they can ignite. Fire prevention is directed toward reducing the escape of vapors, diluting the vapors to a safe level, and eliminating ignition sources such as sparks, static electricity, hot surfaces, or flames.

The use and storage of fluid-preserved scientific specimens in natural history collections is not addressed directly in the Uniform Fire Code or the Uniform Fire Code Standards of the National Fire Prevention Association and International Fire Code Institute. Instead, the fire code addresses the storage of bulk containers of full-strength (95%) alcohol and the storage of retail liquor (usually 5-15% alcohol). The containers and concentrations of fluid preservative used in collections do not fit either of these categories, so it is necessary to work out an acceptable fire prevention and safety plan with the appropriate local officials. Discuss fire and safety issues with local officials so that they understand that (1) fluid preservatives are diluted for museum use; (2) most preserved specimens are kept in small, well-sealed containers; and (3) the risk of spillage or breakage is very low. For more information, see Simmons (2014) and Stemen (2010).

Fluid preservatives may be divided into three classes (I, II, and III) and three groups (A, B, and C) based on their flash point and boiling point. The flash point is the lowest temperature at which a liquid releases

enough vapors to start burning if ignited. Alcohols are flammable liquids; formaldehyde is a combustible liquid. A flammable liquid is defined as one which has a flash point below 100°F (37.8°C). A combustible liquid has a flash point above 100°F (37.8°C). Preservative strength ethyl alcohol (70%) is a Class IB flammable liquid. Preservative strength isopropyl alcohol (40–55%) is a Class IC flammable liquid. The flash points and standard preservative strengths for these fluids are indicated in Table 3.

Fire code interpretations for fluid-preserved collections may include: removing all ignition sources from collection storage areas; limiting shelving to no more than seven feet above the floor; requiring explosion-proof fixtures, electrical outlets, and switches; requiring electrical outlets to be located at least three feet above the floor; requiring fire rated doors, ceilings, walls, and floors; requiring both heat and smoke detectors; and requiring a fire suppression system.

Fire suppression systems may use water (sprinklers or micromist), gas, or foam. Gas and foam systems require space for large storage tanks—the tanks are expensive to fill, and once the gas or foam is exhausted, the suppression system ceases to function. Water-based systems do not require storage tanks, and continue to function as long as there is a supply of water to the building. The most common system in fluid-preserved collections is a sprinkler system. Sprinkler systems in collection storage areas are usually required to have an output of 0.30 gpm/ft², (gallons per minute per square foot) over the entire floor area, and 0.21 gpm/ ft² in laboratory areas, and may require floor drains. Ventilation requirements are designed to prevent the accumulation of vapors. This usually means airflow of 1 ft³/m/ft² (cubic feet per minute per square foot)

Table 3. Commonly used preservatives.

Names	Ethyl alcohol, ethanol, grain alcohol, ETOH	Isopropyl alcohol, isopropanol, 2-propanol, rubbing alcohol	Formaldehyde, formalin
Chemical formula	CH ₃ CH ₂ OH	CH ₃ CHOHCH ₃	HCHO
Flash point	16.6°C in 70% solution	18.3°C in 70% solution	85°C in 3.7% solution
Vapor density (air = 1.0)	1.59	2.07	1.00
Standard preservative strength	70%	Commonly 70%; may be as low as 45%	3.7% ["10% formalin" is 1 part full strength 37% formaldehyde in 9 parts water]
Density of standard preservative at 20°C	0.790 g/mL	0.840 g/mL	1.009 g/mL

for collection storage areas and laboratory ventilation of 5 ft³/m/ft². Restrictions may be imposed on the amount of preservative stored per room. The storage area may require a reduced temperature—a temperature of 65–70°F is preferred because below 65°F, lipids begin to solidify and paraformaldehyde may form in specimen containers (see discussion in Simmons 2014). Access to the storage area may need to be limited and strictly controlled.

Flammable and combustible liquids should not be stored below grade (e.g., in a basement). The Uniform Fire Code limits storage volumes of Class 1B fluids to 15,000 gallons on the ground floor of a building and 12,000 gallons per floor on upper floors. Shelving must have a lip or guard to prevent individual containers from falling. The Uniform Fire Code contains specific requirements for both metal and wooden shelving systems.

When dispensing flammable or combustible liquids from a storage drum, the pump must be grounded to eliminate static electricity and sparking. Connect the supplying container to the receiving container with a bonding wire. Connect a grounding wire between the pump and a grounded structure such as a grounded water pipe or metal framework. A bonding wire or grounding wire is a length of electrical wire with a metal clip on each end for easy attachment to an object or structure. All metal and plastic supply containers, drums, and pumps must be grounded when dispensing preservative.

All storage and laboratory areas should be equipped with spill kits for safe, efficient cleanup of preservative spills. A spill kit is a high-density polyethylene (HDPE) bucket with a lid, containing absorbent pads, a small spill boom, neoprene gloves, safety glasses, a Tyvek suit, and plastic bags (see *Appendix III* for suppliers). Clean up preservative spills immediately with the absorbent pads and booms; enclose the used pads and booms in a plastic bag for disposal.

Laboratory areas should be equipped with eye wash stations and an emergency shower. Eye wash stations should be tested weekly (by flushing water through the nozzle); safety showers should be tested monthly. When working with fluid-preserved specimens or preservative fluids, wear safety glasses and neoprene gloves (which are rated for formaldehyde exposure).

THE PREPARATION LABORATORY

The workspace for specimen processing, cataloging, loan preparation and receipt, and related activities is the preparation laboratory. It should be equipped with bench top fume collectors, a sink with hot and cold water, and cabinets and drawers for the storage of supplies and equipment. It should be well-ventilated, well-lighted, and have UV filters on all ultraviolet producing light sources. A supply of deionized or distilled water should be available for mixing fixative and preservative solutions.

Offices

The offices for collection managers, curators, and researchers should be separate from the preparation laboratory and collection storage areas. Office activities, preparation work, and collection storage are not compatible.

Library

Bibliographic and other reference materials for specimen identification, preservation, and management are essential. The library may also house field notes, collection archives, photographic collections, and other relevant items. The library should be separate from the preparation laboratory and collection storage areas. Preparation activities and collection storage are not compatible with a library environment.

Other Requirements

Catalogs, important documents and records, computers, and electronic storage media should be protected from water, leaks, floods, fire, spilled coffee, and other common museum catastrophes. Catalogs and other important collection records should be backed up by offsite archival microfilm copies (see *Hand-written Catalogs and Electronic Data Storage*). Database records should be backed up regularly and the backup copies stored offsite.

PERSONNEL AND DUTIES

Such staff as there were [at the British Museum] were overworked and underpaid and often had to take on outside work to support themselves. There were no pensions, so appointees tended to stay at their posts until they died or went mad, which they did with astonishing frequency.

—Lynn Barber (1980)

Although the number of personnel will depend upon the size of a particular collection, how it is used, and the institution housing it, the following job titles and general areas of responsibility are found in many museum situations.

Curator

Historically, the curator was the person who had sole responsibility for the care of the collection, but curators now do little actual collection care (Simmons 1993). A natural history museum curator is usually a scientist who conducts specimen-based research. In most museums, the curator has the ultimate responsibility for the collection and exercises intellectual control over the collection, but has minimal involvement in the day-to-day management of the collection (Laub 1985). In addition to research responsibilities, the curator approves requests to use the collection, establishes and maintains taxonomic standards, is responsible for collection growth, and works with the collection manager to establish and implement collection policies. A curator should have a terminal research degree, an expert knowledge of systematic herpetology, and be actively involved in specimen-based research.

Collection Manager

The collection manager manages the collection and performs the collection care duties formerly assigned to the curator (Cato 1991; Ford and Simmons 1997; Simmons 1993, 2015). The collection manager is responsible for the care, conservation, and management of the collection and collection records, and access to specimens and data. The collection manager, with the curator, develops and implements collection policies. The collection manager is

responsible for monitoring the collection storage environment, approving and obtaining collection care materials and chemicals, supervising collection use, and training and supervising other collection care workers. The collection manager makes regular visual inspections of the collection storage areas to detect preservative loss, presence of pests, and disorder in the collection, and records and evaluates collection activities.

A collection manager should be a professional with an undergraduate degree in biology and an advanced degree in museum studies (with particular emphasis on collection management and conservation), an understanding of systematics, and a familiarity with the herpetological literature. Useful skills and knowledge areas for collection managers include technical and grant writing, materials science, collections conservation, storage environments, mechanical ability, familiarity with collection database systems, and information management.

Curatorial Assistants

Curatorial assistants do much of the hands-on work of collection care, working under the direction of the collection manager. Curatorial assistants label containers, tie tags, top off containers of preservative, shelve containers, file documents, pack and unpack specimens, and perform assorted other tasks.

COLLECTION MANAGEMENT

A museum has a responsibility to provide reasonable care of the objects entrusted to it... As a practical matter, this means that ... museum policies and procedures [should] afford prudent care and protection for all museum objects in light of existing circumstances.
—M.C. Malero and I.P. DeAngelis (2012)

Collection Management Policies

The purpose of the collection management policies are to ensure that the institution meets its legal and ethical obligations to care for the collections in a professional manner. The collections management policies should define collection care standards, areas of responsibility, and establish collection documentation requirements. Guidelines for collection management policies are provided by Cato and Williams (1993); Malero and DeAngelis (2012); and Simmons (2006).

The collection management policies should address requirements and standards for accessioning and deaccessioning of specimens, documentation, cataloging, tagging and labeling, standards for the care of collections, access to the collection and use of the collection (including loans, gifts, and exchanges), access to and use of collection information, collection evaluation, and collection growth. These topics are addressed in the appropriate sections below. A guide to locating additional information on collection management policies and procedures is provided in Table 4.

The collection management policies should be reviewed periodically by the curator and the collection manager.

Table 4. Resources for developing collections management policies and procedures.

<i>Policy subject</i>	<i>Section of this publication</i>	<i>Additional references</i>
Field preservation and documentation	Preservation of Specimens	Casas-Andreu et al. 1991; Garrett 1989; Scrocchi and Kretzschmar 1996; Simmons 2014
Conservation of the collection	Conservation of Herpetological Collections	Cato and Williams 1993; National Park Service 1996; Simmons 1995; Society for the Preservation of Natural History Collections 1994
General	Museum Collections; Collections Management Policy	Buck and Gilmore 2010; Cato and Williams 1993; Malaro and DeAngelis 2012; Society for the Preservation of Natural History Collections 1994
Acquisition	The Accession Process; Field Collecting; Guidelines for Collection Growth	Buck and Gilmore 2010; Cato and Williams 1993; Fritts 1976; Lee et al 1982; Malaro and DeAngelis 2012; Merritt and Lidgard 2000
Accession	The Accession Process	Buck and Gilmore 2010; Cato and Williams 1993; Malaro and DeAngelis 2012
Loans	Loans, Gifts, and Exchanges	Buck and Gilmore 2010; Malaro and DeAngelis 2012; Merritt 1992
Collection use	Categories of Loans; Evaluation of Collections	Cato and Williams 1993; Malaro and DeAngelis 2012
Information management	Cataloging Specimens	Buck and Gilmore 2010; Cato and Williams 1993
Destructive and consumptive sampling	Loan Policy	Cato 1993; Cato and Schmidly 1991; Cato and Williams 1993
Deaccessioning	Cataloging Specimens	Cato and Williams 1993; Malaro and DeAngelis 2012
Standards of collections care	Collections Management; Personnel and Duties; The Collection Storage Facility	National Park Service 1996; Simmons 1995; SPNHC 1994
Collection Evaluation	Annual Reports of Collections Management Activities; Evaluation of Collections	McGinley 1993; Merritt and Lidgard 2000; Price and Fitzgerald 1996; Waller 1995; Williams et al. 1996

Accession of Specimens

Accessioning means adding a specimen to the collection. Accessioning is the formal process by which the museum takes legal possession of specimens, and encompasses the entire process from receipt of a specimen in the museum through its preparation for cataloging (Buck and Gilmore 2010, Malaro and DeAngelis 2012, Simmons 2006). It includes the preparation and archiving of appropriate documentation. At the time of accessioning, the specimen data should be checked for completeness and accuracy to avoid delay during the cataloging process.

An accession is one or more objects or specimens received from a single source, at one time. The accession record contains the essential documents and information describing the transaction through which the museum acquired the specimens. It is part of the permanent documentation of the museum. The accession record should show the date of receipt; date of accession; collector's or donor's name, address, postal code, telephone number, and email address; and include a description sufficient to identify the specimens. Each accession is assigned a sequential accession number by the institution. This number should be used to cross-reference all collection documentation and to label the specimens until they are cataloged. The accession number should be listed in the catalog entry for the specimen.

Accessions must be relevant to the purposes and priorities of the collection. The museum must be able to provide proper storage and care of all specimens it accessions without compromising its ability to store and care for what it already has. All accessioned specimens must be obtained legally and they must be properly documented and labeled. The responsibility for the registration of accessions may rest with the collection manager, registrar, curator, or an administrator, depending upon the institutional organization.

Considerations for Accessioning Decisions

1. Do the specimens fall within the categories identified as important to collection growth?
2. Are the specimens appropriate for the collection? Could they be of more use in another institution?
3. Are the specimens well preserved?
4. Are the field notes and other documentation in good order?
5. Are there originals or copies of the necessary permits and declarations for the specimens (e.g., permits to collect, export, and import the specimens, 3-177 declaration, etc.)?
6. Has a clear understanding been reached with the donor regarding the terms of deposit of the specimens (see *Deed of Gift/Deed of Transfer*)?
7. If specimens came from a zoo or other live-animal facility, have proper necropsy protocols been followed (Jacobson 1978)? Were the specimens exposed to toxins, infectious agents, or radiation?

Repository Agreements

A repository agreement is an arrangement whereby a museum takes temporary physical custody of a collection for management purposes. Repository agreements should be time-limited, with a termination date

provided in the documentation, and should specify which institution bears the financial responsibility for returning the collection to the owner upon termination of the agreement. The agreement should specify what limitations, if any, are placed on the managing museum. For example, the managing museum should be able to approve access, loans, and non-destructive use of the specimens on a repository agreement without consultation with the owner of the collection, but should get permission of the owner before invasive procedures are conducted or ownership of any specimens is transferred to another institution. Most repository agreements are long-term (e.g., 20 years is common) with a provision for renewal at the termination of the agreement. It is recommended that documentation for a repository agreement parallel the normal accessioning and registration procedures in the museum as much as possible. Repository agreements have been standard for many years in archaeological collections, but are a fairly new type of arrangement in herpetological collections.

Documentation of Collections

Field Notes

The field notes consist of the field journal and the field catalog prepared by the collector at the time the specimens were collected in the field (see *Field Notes*). The field journal and field notes are extremely valuable. These documents should be archived in the same institution as the specimens, and cross-referenced to include the museum catalog numbers assigned to the specimens. If it is not possible to obtain the original field notes, at least obtain an archival quality copy of them.

Legal Documents

All incoming specimens must be obtained legally (see *Permits*). Originals or archival quality copies of all permits, declarations, export and import forms, other permissions to obtain, possess, and transport the specimens, correspondence, and deed of gift or deed of transfer are part of the accession record.

Images

Photographs, negatives, color transparencies, and digital images should be referenced with the accession number and, where appropriate, with the museum catalog number of the individual specimen(s) depicted. Each image should be labeled with a separate, sequential number. Store images using appropriate archival quality materials (see *The Storage Environment for Herpetological Collections*).

Audio and Video Recordings

Each recording should be labeled with a separate, sequential number. Audio and video recordings on tape have a very limited life. Tape must be handled with great care. The only way to prolong the useful life of tape recordings is to digitize the recorded information and store it in a medium acceptable for longer-term storage (e.g., on compact disc).

The Accessioning Process

When specimens are received, they should be unpacked, properly housed, and labeled pending accession.

Step 1. Receiving the Specimens

Inspect the container that the specimens arrive in for any damage or signs of leakage or tampering. Take photographs of the container if any potential problems are detected. Unpack the container with care, watching for loose specimens, ruptured containers, information written on containers, field notes stuck between the packed specimens, or slips of paper containing data tucked in with specimens but not attached to them. Some collectors can be quite creative when they pack specimens. Check all specimens against the packing list provided. If no such list was provided, make a complete list of specimens received and include it in the accession documentation. Provisionally mark each container with the name of the source (e.g., donor or collector of the specimens) until an accession number becomes available.

Step 2. Complete an Accession Checklist

Complete an accession checklist for the specimens. The accession checklist is a standardized form that is used for each potential accession to help ensure that all necessary documentation is in place for the museum to assume ownership of the collection. An example of an accession checklist is shown in Fig. 17. The accession checklist should include:

- *Accession number* (to be filled in later when it is assigned by the museum).
- *Catalog numbers* when they are assigned to the specimens.
- *Collectors*. Full names of all collectors who contributed to the collection.
- *Date* the specimens were received by the museum.
- *Description* of the collection, including the number of individual specimens
- *List of permits, field notes, correspondence, and other documentation* that accompanies the collection.
- *Name and signature* of the person responsible for completing the accession checklist.
- *Source*. Name, address, postal code, telephone, and email address of the collector, donor, seller, or museum donating the specimens.

Step 3. Assemble the Accession File

Assemble the documents that make up the Accession File. These include:

- Accession Checklist.
- Correspondence relating to the collection, transport, and ownership of the specimens.
- Deed of Gift/Deed of Transfer.
- Field Notes (these are not usually physically included in the file itself, but their location in the museum should be noted).
- Permits.
- Other documents pertaining to the accession.

Step 4. Obtain an Accession Number

Submit the appropriate documentation to obtain an accession number (see *Accession of Specimens*). This step establishes the legality of the accession,

Accession Checklist

The following checklist is to be completed before any specimen or object may be accessioned by the Natural History Museum. Check boxes ONLY if the required documents are in hand.

RECEIPT OF SPECIMENS PREVIOUSLY CATALOGED IN ANOTHER MUSEUM (by donation, exchange, gift, abandonment, or purchase)	Yes	No	Not Applicable
Transmittal form or letter from appropriate authority at the institution of origin	☐	☐	☐
Signed Deed of Transfer form	☐	☐	☐
Export permit (if from a non-US institution)	☐	☐	☐
Import permit (if from a non-US institution)	☐	☐	☐
CITES permits if transaction involves CITES-listed specimens	☐	☐	☐
APHIS certification	☐	☐	☐
Other:	☐	☐	☐

RECEIPT OF SPECIMENS NOT PREVIOUSLY CATALOGED IN ANOTHER MUSEUM (field work, gift, exchange, purchase, donation, bequest, or contract)	Yes	No	Not Applicable
Original or copy of collecting permit(s)	☐	☐	☐
Original or copy of export permit (if from a non-US institution)	☐	☐	☐
Signed Deed of Transfer form	☐	☐	☐
Current US Fish and Wildlife Service permit on file	☐	☐	☐
Copy of 3-177 form (if from a non-US institution)	☐	☐	☐
CITES permits if transaction involves CITES-listed specimens	☐	☐	☐
Antarctic Conservation Act permit	☐	☐	☐
Bald Eagle Protection Act permit	☐	☐	☐
Bureau of Land Management permit	☐	☐	☐
Controlled Substances Act	☐	☐	☐
Feather Import Quota	☐	☐	☐
Federal Noxious Weed Act	☐	☐	☐
Fur Seal Act	☐	☐	☐
Marine Mammal Protection Act permit	☐	☐	☐
Migratory Bird Treaty Act permit	☐	☐	☐
Plant Pest Act	☐	☐	☐
Plant Quarantine Act	☐	☐	☐
State Collecting permit	☐	☐	☐
US Fish and Wildlife Salvage permit	☐	☐	☐
APHIS certification	☐	☐	☐
Original or copy of field notes for specimens in the accession	☐	☐	☐
Originals or copies of any correspondence relating to this accession	☐	☐	☐
Other:	☐	☐	☐

Figure 17. Accession checklist.

I hereby certify that to the best of my knowledge the above information is correct and accurate and that the specimens and/or objects comprising this accession were obtained legally. Signature: _____ Title: _____ Date: _____		
This accession is approved for acceptance and cataloging by the Natural History Museum of the University of Kansas. Signature: _____ Title: _____ Date: _____		
Accession Number: _____		
Catalog numbers assigned to accession:		
List of support documents:		

Figure 17. Accession checklist (continued).

and is the point at which the museum takes legal ownership of the specimens. Label all documents and specimen containers with the accession number as soon as the number is available.

Deed of Gift/Transfer

Each donating institution or individual must sign a Deed of Gift or Deed of Transfer document (Malaro and DeAngelis 2012). This document (1) is a statement of the good-faith effort of the institution or individual to demonstrate that the accession was obtained legally; and (2) establishes the museum's clear ownership of the specimens. An example of a Deed of Transfer is shown in Fig. 18.

Selection of a Preservative

Since the mid-1600s, when the preservative properties of spirits of wine were discovered, the overwhelmingly preferred fluid preservative has been ethyl alcohol, usually in a 60–75% concentration (Simmons 2014). The other common preservatives (though used far less than ethyl alcohol) are isopropyl alcohol and formaldehyde. Isopropyl alcohol was not used as a preservative until sometime after 1920 when commercial production of it began (Hatch

NATURAL HISTORY MUSEUM Deed of Transfer	
Date: _____	
Received from: Name: _____ Address: _____ _____ _____	
Telephone: _____	
Description of Accession:	
AGREEMENT	
I hereby acknowledge that I have read the terms of acceptance of this accession, and that to the best of my knowledge, the specimens and/or objects comprising this accession were obtained legally. Signature of Donor or Agent: _____ Date: _____	

Figure 18. Deed of gift/transfer.

1961); formaldehyde was not used as a preservative until sometime after 1893 (Simmons 2014).

Isopropyl alcohol does not have the 350+ year record of use in museums that ethyl alcohol does. The biggest drawbacks to ethyl alcohol are its expense, the necessity of obtaining a federal permit to purchase it tax-free, the amount of shrinkage specimens preserved in it undergo, and the potential fire hazard it poses. The arguments in favor of using isopropyl alcohol are that it is less expensive and easier to obtain (a federal permit is not required), and that it leaves the specimens more flexible (Walker et al. 1995). However, isopropyl alcohol also poses a fire hazard, it is twice as toxic as ethyl alcohol, many people find its odor objectionable, and it has been shown to cause greater shrinkage of specimens than ethanol (see discussion in Simmons 2014). According to Stoddard (1989), the greater flexibility of specimens preserved in isopropyl is due to more extensive breakdown of the tissue matrix compared to specimens preserved in ethanol; the greater flexibility of specimens may mask warning signs of deterioration. Furthermore, the use of isopropyl alcohol as a preservative makes specimens unsuitable for most types of histological preparation (Jones and Owen 1987). It can be difficult to measure the density of isopropyl alcohol solutions when using a hydrometer because its density is close to that of water. In low concentrations (below 45%), isopropyl may promote a rapid clearing of tissues. Some workers have reported that isopropyl alcohol is difficult to mix with water and prone to layering in containers (Fink et al. 1979). In long-term storage, it has been shown to soften bone (Steedman 1976). While an argument can be made to continue using isopropyl for fish specimens already in it (Walker et al. 1995), this argument does not justify using a preservative other than ethyl alcohol to preserve herpetological collections.

Just because most fluid-preserved collections use ethyl alcohol does not mean that it is the best preservative to use; however, no data indicate that any other preservative is superior, or even the equal of ethyl alcohol (Simmons 2014). Some alternate preservative formulas have been proposed for specific functions, such as preserving colors of specimens (see *Color Preservation*), or making specimens useful for specific histological techniques, and a set of criteria for a new fluid preservative has been proposed (van Dam 2003). In the absence of carefully controlled, long-term or accelerated aging studies of other preservative solutions to demonstrate their superiority, fluid-preserved specimens should continue to be kept in ethyl alcohol.

Transfer of Specimens from Fixative to Preservative

It has long been the custom to remove specimens from the field fixative, rinse or soak them in water in open containers for one or more days, and then place them directly in storage-strength alcohol preservative (Simmons 1995). There are several problems with this procedure: (1) as the formaldehyde is removed by the water, enzymatic activity in the tissues may resume (Taylor 1981a); (2) as the formaldehyde crosslinks are removed, autolysis may occur; (3) specimens will be stressed as they swell (with the absorption of water) and shrink once they are placed in alcohol (as they dehydrate); and (4) the water extracted from the specimens will dilute the preservative solution they are placed in. To avoid these problems, some workers have suggested transferring

specimens directly from the formaldehyde fixative to the alcohol preservative (Fink et al. 1979), but this procedure also has several drawbacks, including (1) severe osmotic shock in going from nearly 100% water to 30% water, and (2) the quantity of formaldehyde that is released into the alcohol preservative will drive the acidification of the alcohol solution, which may decalcify specimens and cause more severe color changes, as well as pose serious health risks for people working with the specimens. Based on the studies available on shrinkage, swelling, and other effects on specimens during transfer (e.g., Jones and Owen 1987; Laframboise et al. 1993; see discussion in Simmons 2014), and considering the effects of osmotic pressure changes on tissues (Steedman 1976), a more gradual transfer is recommended. Because osmotic pressure rises steadily with ethanol concentrations below about 75%, it has been suggested that approximately equal concentration increments are the most appropriate for stepping specimens up to higher ethanol concentrations (Waller and Strang 1996). Following the protocol of Laframboise et al. (1993), I recommend using steps of approximately 20% to stage specimens from 10% formalin solutions to 70%, ethyl alcohol (Fig. 19).

Trace Amounts of Formaldehyde in Preservative Solutions

It is difficult (and probably undesirable) to completely remove the formaldehyde fixative from specimens fixed in 10% formalin by washing (Simmons 2014). As a result, some trace amounts of formaldehyde will remain in the alcohol preservative. One study found these amounts to average 0.07% in one collection (Waller and Simmons 2003). Trace amounts of formaldehyde in preservative solutions can be detected by using formaldehyde test strips, which can be made by following the instructions in Waller and McAlister (1996), or a commercial formaldehyde test kit may be purchased (see *Appendix III*). Trace amounts of formaldehyde are usually too small to affect the quality of the preservative; however, in some cases formaldehyde and other sources of acid (e.g., label paper) have been shown to acidify alcohol solutions (Andrei and Genoways 1999; Van Guelpen 1999). Because the trace amounts of formaldehyde and other chemicals may be present in alcohol preservatives, it is recommended that neoprene gloves be worn when handling any fluid-preserved specimen.

Transfer of Specimens between Alcohols

As a general rule, specimens should not be changed from one type of alcohol to another, especially specimens on loan from other institutions. Changing a specimen from one type of alcohol to another may cause significant shrinkage and other damage that could significantly alter the body proportions of the specimens (see review in Simmons 2014). If it is necessary to change

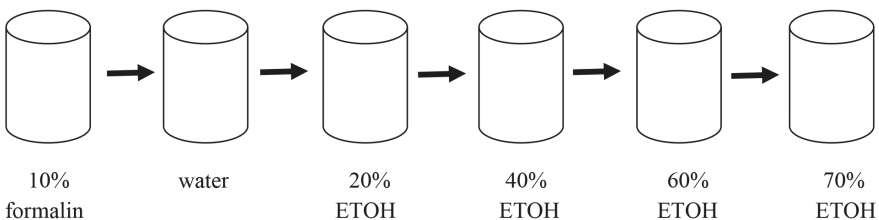


Figure 19. Transfer of specimens from field fixative to museum preservative.

specimens from one alcohol preservative to another, follow the protocol of Laframboise et al. (1993).

CATALOGING SPECIMENS

Cataloging is the process by which a specimen is assigned a permanent reference number. The catalog number associates the field data with the specimen, makes it possible to retrieve the specimen from the collection, and identifies the specimen for all subsequent uses in the collection. The catalog number is not assigned to a specimen until it has been through the complete accession process (see *Accession of Specimens*), because catalog numbers may not be assigned to specimens until the museum can demonstrate its ownership of them (there is one exception to this rule—under the terms of some repository agreements, specimens in temporary possession of the museum are cataloged for management purposes. See *Repository Agreements* for further discussion).

Catalog numbers of deaccessioned or destroyed specimens should never be reassigned to other specimens.

Cross-cataloging refers to the secondary sorting of specimen data into other categories (e.g., by geography or by taxa). Historically, this procedure required the creation of new hard-copy specimen records, but now cross-cataloging is most efficiently done with an electronic database.

Hand-Written Catalogs and Electronic Data Storage

For hundreds of years, the tradition in natural history museums has been to maintain a hand-written, sequential catalog (also called a register) that lists each individual specimen by number, along with pertinent data. It is very difficult to work with the information in this format, so most institutions also maintained card files or loose-leaf notebooks in which the information was rearranged by categories (see *Cross-cataloging*). Now that specimen data may be more easily and efficiently manipulated in an electronic database (see *Electronic Databases*), many institutions have ceased to maintain their hand-written catalogs, usually citing the presumed expense of writing information by hand. Although an electronic database is vastly superior for data manipulation, electronic data are not permanent. It is important to distinguish between an archival copy of catalog data and a backup copy. An archival copy is a permanent document, made using the best materials available for long-term storage. A backup copy of an electronic database is a safeguard used to rebuild the database should loss occur. Electronic backup copies are not archival (see discussion below) although a good microfilm copy of a hand-written catalog can be.

No system of recording data is as permanent as using an archival black ink on acid-free 100% rag paper. If cared for properly, a hand-written catalog on good paper will last at least 400 years, probably much longer, and still be legible to a reader. No form of electronic data storage or machine printing will last this long, much less be readable long into the future (Baca, Coburn, and Hubbard 2008; Simmons 1998). The only long-term means of storing digitized data at present are compact disks (CD), external hard drives or flash drives, or cloud storage (the use of logical data pools

on multiple servers). CDs are made of a polycarbonate compound that is more susceptible to damage from heat, moisture, light (which causes it to discolor), abrasion, and chemical exposure (especially to compounds containing benzene rings, such as naphthalene or paradichlorobenzene) than is paper. A CD also requires a functioning CD reader for access to the data. External hard drives, flash drives, and similar devices depend on magnetically charged metallic particles that are susceptible to data loss from static electricity, humidity, high temperature, physical and electric shocks, mechanical failure, the deterioration of the material that the particles are embedded in, and also require functioning hardware and software. Cloud storage utilizes multiple servers (that the user has no control over) to store information in media similar to hard drives. It is often assumed that digitally stored data can simply be accurately transferred from one generation of digital storage to the next, however, data transfer always results in some loss and/or corruption of data, and it is not always possible to transfer stored electronic information between generations of hardware. Preserving electronic data obligates the museum to a perpetual expense in data transfer as software is updated and technology transfer as each new generation of computers appears. Upgrades should not be skipped, as failure to upgrade increases the risk of later data loss. Multiple backups will reduce the risk of data loss during data transfer. Database maintenance should not interfere with the collection managers' collection care duties. The primary focus of the collection manager should be care of the collection, with the database as just one of many tools employed for that purpose.

Despite the vast quantity of information being recorded in digital formats today, at this time, electronic data in a digital format is not archival (Hedstrom 1998). As one group of experts stated, "The notion that once something has become digital it has somehow been made permanent lingers, but in fact the strength of digital files is in such things as their transportability, or the ease with which they can be processed by computer or reproduced—and they are actually likely to face an early death" (Baca et al. 2008:120–121).

Although maintaining an electronic database is extremely important, abandoning the hand-written catalog is foolish. The hand-written listing of specimens added to the collection is a legal document that records the sequence in which objects were added to the collection (Baron 1991). Unlike electronic databases, records cannot be altered or eliminated from a hand-written ledger without leaving behind evidence of the modification. Electronic databases require regular backup, regular software upgrades, and periodic hardware upgrades, but still do not provide the permanent association of specimens and catalog numbers that a handwritten catalog provides. Few electronic databases preserve the specimen record as originally written, yet this information is often critical in evaluating a specimen record years after it is entered in the database. Hand-written catalogs are expensive to buy and take time to write in, but they remain the only permanent, long-term means of meeting the museum's legal obligation to preserve vital collection data.

Paper Catalog Books

The hand-written catalog should contain pen-ruled sheets of acid free, 100% rag paper, folded in signatures and permanently bound in hard covers. Because the hand-written catalog is the primary listing of specimens in the collection, it must be treated as an archival document and carefully protected. A preservation quality microfilm backup copy should be stored off-site.

Hand-Written Catalog Entries

Information in the hand-written catalog should be neatly printed in an archivally stable black ink with a technical pen (see discussion of inks in *Field Notes*). Minor mistakes may be neatly erased with a clean, high quality ink eraser; larger errors should be crossed out with one line, the correction written in above, then initialed by the cataloger.

Electronic Databases

There are a wide variety of electronic database programs on the market that are suitable for use with herpetological collections. Which database is best will depend, in part, on the size and complexity of the collection, how it is used, and the ability of the database to interface with online data sharing sites such as VertNet.

Collections management systems vary from products such as KE EMu (Sandino 2009) and SPECIFY that are designed specifically for natural history collections to open source software used by object collectors of all sorts. The selection of a comprehensive collections management system is highly recommended. A comprehensive system is a relational database that provides customizable templates for collections tasks (e.g., data entry, loan forms, deeds of gift, shipping invoices), has many data fields, has the ability to retrieve information based on any data field, has a built-in report writer, interfaces for multiple backup systems, allows correlation of support documentation (such as field notes, audio files, and image files), accommodates ancillary information such as references to citations of individual specimens by catalog number in the literature, and is expandable. Inexpensive spreadsheets or systems that do not support relationships between tables and require extensive user input to achieve functionality (e.g., Excel, Foxpro) are not recommended—at best, such systems can be made to duplicate an old-fashioned paper system, but will not enhance the management of the collection (Simmons 2013).

The Cataloging Process

Once a collection has been accessioned, specimen identification should be confirmed, collecting locality information standardized, and the specimens assigned catalog numbers. Whether making entries in a hand-written catalog or using an electronic database, the process should be based on the following general outline. Some electronic databases allow the collectors themselves to prepare electronic data files for their specimens which can be transferred into a collection database—before this is done, entries should be verified with the collector's original field notes and data fields should be checked for correct format and syntax.

Arrangement of Specimens for Cataloging

The specimens to be cataloged should be arranged so that within a given locality, all individuals of each species can be numbered sequentially. This method enables the cataloging process to proceed much faster and more efficiently by maximizing amount of data that can be duplicated, reduces error rates, saves space by allowing series to be printed on container labels (Williams et al. 1977), and makes it easier to locate individual specimens of a geographic or taxonomic unit.

The following information should be included in the catalog record for each specimen.

Standard Locality. Localities should be described so that they are easy to locate on maps, and should be specific enough to eliminate any ambiguities for subsequent users of the data. Refer to Riemer (1954), Axtell (1965), and Hutchinson (1964) for a discussion of map localities and standardized formats.

GPS Localities and Standard Geographic Coordinates. Include GPS (global positioning system) locality data or standard geographic coordinates whenever possible. For information on georeferencing localities for which GPS data from the field are not available, see Graham et al. (2006); Guralnick et al. (2006); Murphey et al. (2004); and van Erp et al. (2014).

The traditional catalog entry in standard format is written as follows:

Country: State, Province, District, or Region: Other subdivision(s), as appropriate: Distance and direction to nearest map locality, coordinates, elevation

Example:

Brazil: Amazonas: Castanho: approximately 40 km S Manaus, km 12 on road to Autazes, 03° 37' 10.4" S, 59° 86' 78.4" W, 50 m

Date. Enter dates as the numerical day, the first three letters of the month, and the full numerical year, as in 12 Feb 1809. Systems using all numerals are easily misunderstood, because the convention determining whether day or month comes first varies from country to country.

Scientific Name. Personnel at each institution must determine which taxonomic reference lists they will follow. Consistent use of a standard taxonomy is recommended; the curator and collection manager must make judgments concerning names they will consider valid. Useful on-line taxonomic sources are listed in *Appendix III*.

Collector. Collector(s) full name(s) should be recorded in the accession documents. Last names and initials may be used in the catalog record.

Field Number or Original Number. Record the field number for a specimen received directly from the collector, or the original museum catalog number

for a specimen previously cataloged in another collection. Museum names may be abbreviated by using the standard symbolic code of the institution (Leviton et al. 1985; see also *Appendix III*).

Preparation Type. The default for a herpetological collection is a whole adult specimen in an alcohol preservative. Note in the specimen record if the specimen is prepared as other than a standard preparation (e.g., a larvae, dry skeleton, cleared and stained preparation).

Storage Medium. Note if the fluid preservative is something other than the standard preservative in use (e.g., Bouin's solution, glycerin), or if the specimens must be stored under special conditions (e.g., dry).

Type Status. Note if the specimen is a holotype or paratype of a valid or synonymized taxon.

Developmental Stage of Specimen. The default is a sexually mature adult. Note if the specimen is a juvenile, neonate, egg, larvae, etc.

Accession Number. Record the accession number the specimen is associated with.

Remarks

- Record any reference(s) to any specimen documentation such as images, recordings, field notes, or other pertinent documentation.
- Note if the specimen was maintained live in captivity before it was processed, if it was captive born, or captive raised.
- Record other information as appropriate.

Tagging Specimens

Once a specimen has been assigned a catalog number, attach the numbered tag to the specimen (see *Specimen Tags*).

1. Tie the tag on the specimen's left hind leg, just below the constriction of the knee, firmly but not tight enough to damage the specimen. The collector's field tag and the museum tag should be placed in the same position on the specimen.
2. If the specimen's left hind leg is missing or damaged, tie the tag on the right hind leg, an arm, or around the specimen's waist, as appropriate.
3. For caecilians, limbless lizards and salamanders, amphisbaenians, and small snakes, tie the tag around the neck if there is sufficient constriction of the neck to keep the tag from coming off. Sew the tag on larger specimens or those lacking a suitable constriction of the neck. Sew through the neck below the vertebral column, or through the skin of the neck, with a thin needle.
4. For animals with limbs too small or too fragile for leg tags, tie the tag around the animal's waist.
5. To associate numbered tags with specimens too small or too fragile for a waist tie, place the individual specimen and the tag (with about an inch of string attached) in a shell vial filled with the appropriate fluid preservative. Position the specimen and tag in the vial so that the tag can be read from the outside. Close the vial with a secure plug of polyester fiber. Avoid leaving bubbles below the polyester plug. Place

the shell vial in a standard size jar (with closure) also filled with the appropriate fluid preservative.

6. For catalog numbers assigned to larvae (whether one individual or a lot), place the tag (with about an inch of string attached) in a shell vial with the specimen(s), and fill it with the appropriate fluid preservative. Position the tag so that it can be read from the outside. Close the vial with a secure plug of polyester fiber. Avoid leaving bubbles below the polyester plug. Place the shell vial in a standard size jar (with closure) also filled with the appropriate fluid preservative.
7. Never remove field tags or previous museum tags from a specimen unless the tags are deteriorating. Deteriorating tags should be replaced with substitutes, and the originals archived in Mylar enclosures (Kishinami 1989, 1992).

Specimen Tags

Historically, specimen tags have been almost as varied as the specimens that they were attached to (Hawks and Williams 1986). Tags have been made of paper, wood, metal, plastic, and cloth, and inscribed with everything from simple numbers to complete collecting and locality data. For use with fluid-preserved specimens, tags should be kept small and simple.

1. Use a small tag format to reduce the possibility of damage to the tag or specimen.
2. Choose a fluid-resistant material that is durable but does not have sharp edges or corners that could damage specimens.
3. Do not use materials that might damage the specimen as they deteriorate (e.g., polyvinylchloride plastic).
4. The tag should have the museum name, acronym, or symbolic code (Leviton et al. 1985; see also *Appendix III*), plus the catalog number on it.

Preferred tag materials include white, sturdy, 100% cotton stock, with a pH of 6.5 to 7.0 (Hawks and Williams 1986), machine printed in indelible carbon black printing ink, or spunbonded polyethylene (sold under such names as Tyvek or Polypaper or for use with thermal transfer printers, as discussed below). A small hole should be punched in one end of the tag to accommodate a soft thread for attaching the tag to the specimen. Do not use plastic tag stock made of unstable materials, plastics with added color, or materials with adhesive backing (e.g., Dymo labels) because all of these materials will contaminate preservative solutions.

Long-lasting, easy to read tags and labels can be printed with a thermal transfer printer (Bentley 2004). A good thermal printer will use a carbon-coated ribbon, print very small letters and numbers clearly on a spun-bonded polyester substrate that meets ASTM standards (Bentley 2004), and communicate with the database.

Stringing Specimen Tags

A tag vice facilitates stringing tags quickly and efficiently. A tag vice is made from two lengths of wood, 12-15 inches long, which are clamped together by two bolts (Fig. 20). A sheet of tags is clamped in the vice with the holes outward for stringing. The vice holds the tags rigid and frees both hands for stringing. Tags should be strung with soft cotton or linen thread so that they

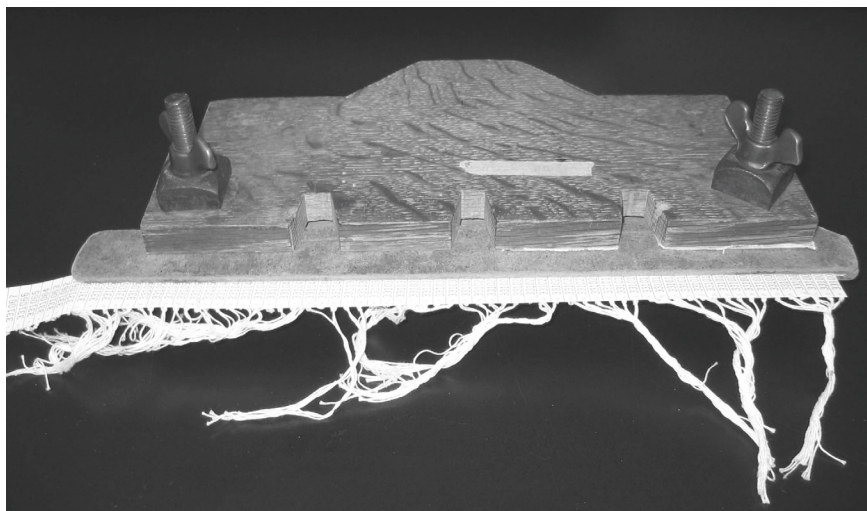


Figure 20. Tag vice for holding tags while attaching string.

are ready to attach to the specimen (I prefer a 100% mercerized soft crochet thread without dye).

Preparation of Labels

Each tagged specimen in the collection is kept in a container (jar, vial, tank, bucket, box, etc.) with a label bearing the specimen's catalog number, scientific name, and locality. Standard labels are printed on fluid-resistant, durable stock. The preferred materials for container labels are spun bonded polyethylene printed with a thermal printer (see discussion above) or white, 100% cotton stock, with a pH of 6.5 to 7.0 (Hawks and Williams 1986). An alternative to machine produced labels for fluid collections are labels that are handwritten in indelible ink with a technical pen. Allow ink to dry for 24 hr before submerging in liquid. Always test each combination of paper and ink before using it in the collection.

One popular label stock used in fluid collections, Resistal paper, is acidic and should not be used. It is fluid resistant due to a melamine ($C_3H_6N_6$) coating, which causes the paper to acidify preservative solutions (Andrei and Genoways 1999; Van Guelpen 1999).

Combining Specimens in Containers

To save space and container costs, multiple specimens of the same species from different localities are often combined in the same container. The drawbacks to this practice include the possibility that smaller or fragile specimens may be damaged by larger specimens or by crowding in the container, and that more specimens must be handled to locate the specimen that is needed, but these issues can be addressed by judicious selection of specimens to combine. A potentially more serious drawback is that when a specimen sits in a container of alcohol, lipids and proteins leach out of the specimen into the solvent, making the surrounding preservative part of the

specimen, too (Von Endt 1994), and mixing specimens in containers means mixing the extracted constituents in the fluid.

When combining specimens in containers, put like-with-like. Do not mix different species in the same container, and do not put too many specimens in one container. When allocating specimens to containers, maintain a ratio of at least 7:3 of volume of liquid to volume of specimens (Simmons 2014).

Container Labels

The standard label for a container should show the following information:

- **Catalog number** of each specimen in the container. List multiple numbers in sequential order.
- **Scientific name** of the species in the container.
- **Family** that the genus and species is in.
- **Locality** in standard format.
- **Type status** of the specimen, if any.
- **Preservative fluid** in the container, if other than the standard default fluid preservative in the collection.
- Note if the specimen should remain dry, or if the container is an empty jar marking the spot for a specimen in a tank.

CONTAINERS AND CLOSURES

A container is a receptacle that holds a specimen and the preservative fluid (see Simmons 1995, 2014). Containers are usually jars, buckets, or tanks (a bottle has a much greater constriction of the neck and mouth than a jar). A closure (e.g., a lid and gasket) is the means of sealing the mouth of the container. Closures are either lids or stoppers (Fig. 21). A lid fits on top of a container, or over the top of a container opening. A stopper fits inside a container opening.

General container types are summarized in Table 5. The best containers currently on the market are the alcohol-resistant, precision-ground glass-stoppered museum jars made of borosilicate glass (Clark 1992; 1993). Considering price, availability, and serviceability, the most suitable containers on the market are clear flint glass jars, with flexible polypropylene threaded lids, with liners or inserts (Suzumoto 1992b).

Glass Containers

Most fluid-preserved specimens are housed in glass jars. Most glass is susceptible to slow deterioration from alcohol and other preservatives. Older museum jars often look dirty or exhibit iridescence when dry, but when wet appear normal—this is the result of glass deterioration. The deterioration is a slow process, but over time the glass becomes fragile and the deterioration products contaminate the preservative fluid and specimens. Glass deterioration is accelerated by repeated wetting and drying of the glassware, by exposure to high relative humidity, or by storage at warm temperatures (Simmons 2014). Borosilicate glass is resistant to alcohol and has excellent optical properties, but it is much more expensive than soda-lime glass. Precision-made borosilicate jars with glass stoppers are now available for fluid-preserved specimens (see *Appendix III*). Although expensive, these jars may be a good alternative for specimens used for exhibit purposes.

Older glassware should be monitored closely for signs of deterioration.

Closures

There are five basic types of closures used for containers of fluid-preserved specimens:

1. Compressible stoppers
2. Non-compressible stoppers
3. Lids held in place by a wire bail or clamp
4. Snap-on plastic lids
5. Threaded lids

Closures are often equipped with a liner, a gasket, or an insert to improve the seal (Fig. 21). A liner is a flat disk that fits inside the lid, usually made of polyethylene or Teflon. A gasket is a compressible material formed in a shape to fit around the rim of the container or the lid. Gaskets may be made of natural rubber (which is usually red or orange), buna-n, buna-n duro (a 50/50 acrylonitrile-butadiene copolymer), or EPDM (ethylene-propylene-diene terpolymer). An insert is a plastic device made to fit into the mouth of the container so that it is held in place when the closure is applied.

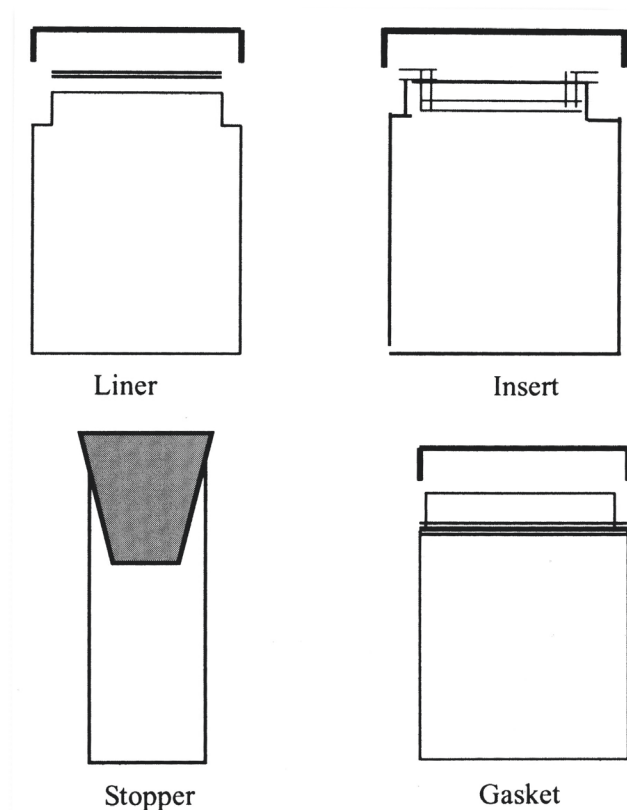


Figure 21. Closures and seals.

Table 5. General container types.

Type of container	Relative cost	Availability	Quality of seal	Longevity of seal	Rating for use
Glass jar with glass stopper ¹	Very high	Poor	Excellent	Life of container	Excellent
Glass jar with glass lid and wire bail	High	Poor	Fair to good	<10 years	Good
Glass jar with thermoplastic threaded lid (polypropylene)	Medium	Very good	Very good	>20 years	Very good
Glass jar with snap-on lid (polyethylene) ²	Medium	Poor	Fair	5-10 years	Poor
Glass jar with thermoset threaded lid (e.g., phenol, melamine, Bakelite)	Medium	Good	Fair	<5 years	Poor
Glass vial with compressible stopper (cork, rubber, synthetic polymer)	Low	Good	Fair	<10 years	Poor
Polyethylene terephthalate (PET) container with polypropylene lid	Low	Fair	Unknown	Unknown	Unknown
Polycarbonate container	Medium	Fair	Fair to good	<10 years	Fair
Ceramic crock	Medium	Poor	Poor	<20 years	Poor
Stainless steel tank	Very high	Good	Fair to good	<10 years ³	Fair
High-density polyethylene (HDPE) bucket or barrel	Low	Good	Good	>15 years	Good

¹Rating is for new borosilicate jars only, not vintage glass jars with glass stoppers.²Reliable snap-on lids available in Europe ("Copenhagen jars") but not in the US.³Seal longevity depends on contact with preservative and extent of compression by lid.

Compressible Stoppers

Compressible stoppers may be made of cork, rubber, or a synthetic material (the most common synthetic stopper material is neoprene). All compressible stoppers lose their elasticity and become brittle as they age, and from contact with the preservative (Fig. 22). When a stopper fails, evaporation occurs, and the preservative is contaminated by the components of the stopper that leach into it. The result is usually dehydrated and discolored specimens and labels (Nizinski 1987). Cork stoppers contaminate the preservative with tannins and pigments, and crack with age (Galigher and Kozloff 1971; Levi 1966).

Non-Compressible Stoppers

Non-compressible stoppers are usually made of glass. These are simple closures (no gasket), but petroleum jelly, oil, or a jar cement may be used to enhance the seal. The old style glass stoppers are not interchangeable (they were individually ground to fit a particular jar) and can be difficult to remove from the jars. The newer borosilicate jars have interchangeable, precision-ground stoppers with an excellent seal. Although very expensive, they may be cost-effective in a large collection in which containers are not opened frequently, and they are very useful for exhibition purposes.

Lids Held in Place by a Clamp or Bail

Some jars have glass lids held fast by pressure from a wire bail or wire clamp, with a compressible gasket to complete the seal. As the gasket ages, it leaches chemicals that contaminate and discolor the preservative. Over time, the gasket loses its elasticity and may crack or liquefy (depending on its composition),



Figure 22. Distortion of compressible stoppers.

so that it can no longer maintain a seal against evaporation. Gaskets made of synthetic material are more durable than those made of natural rubber, but still susceptible to deterioration. Because reliable replacement gaskets are becoming increasingly difficult to find, and because gasket deterioration may contaminate specimens, it is strongly recommended that wire bail jars be phased out of collection use. Recently manufactured glass top bail jars rarely seal well (they are made for storage of dry materials, not fluids).

Snap-On Lids

The snap-on, flexible, polyethylene lids available in the United States usually crack and fail within a year or two due to the degradation of the plastic from age or from exposure to ultraviolet light (e.g., from overhead fluorescent light sources). However, similar closures available in many European countries (locally called Copenhagen jars and lids) are much more reliable.

Threaded Lids

Threaded (screw-on) jar lids may be made of metal or either flexible or rigid plastic. Metal lids are prone to oxidation, and should be avoided if suitable alternatives are available. If metal lids are used, they should be closely monitored for signs of oxidation (Warén et al. 2010).

Rigid thermoset lids (e.g., hard, black Bakelite lids) back-off or crack with fluctuations in temperature. Bakelite lids are prone to embrittlement, particularly in the presence of formaldehyde vapors. Similar but more durable rigid lids made of other phenolic resins are available with a polytetrafluoroethylene (Teflon) liner, sold as PHEN-PTFE closures (see Appendix III). Rigid lids back-off (unscrew themselves) due to differences in thermal expansion between the glass jar and the lid material when subjected to fluctuations in temperature. A 2°C daily change in temperature in the collection storage room can cause a lid to back-off by half-a-turn in just three years (R. Waller, pers. comm.).

Thermoplastic polypropylene lids are flexible enough to provide a good seal, but durable enough to be long-lasting in collection use. I recommend using a polypropylene lid with a knurled edge and a Teflon liner.

Screw-on lids must have continuous threads. Screw-on lids with gaps in the threads do not provide an adequate seal to prevent the evaporation of fluid preservatives.

How to Improve Closure Seals

There are several ways that the seals on glass jars with threaded lids might be improved if you cannot afford to replace the closures:

1. Use a polyethylene liner or insert between the lid and the jar.
2. Wrap the jar and lid junction with polypropylene/acrylic (PPA) adhesive transparent tape (Steigerwald and LaFramboise 1994, 1996) (see *Appendix III*).
3. Wrap the jar threads with Teflon pipe joint tape before applying the lid.
4. Use a sheet of Teflon, polyethylene film, or Parafilm between the container and the closure (see *Appendix III*).
5. Coat the jar threads with petroleum jelly (e.g., Vaseline).

Plastic Containers

It is possible that clear polyethylene terephthalate (PET) containers may provide a low-cost alternative to glass containers (Walker et al. 1999). At present the variety and availability of these jars is limited, most of the jars on the market do not have closures that are adequate to prevent evaporation, and PET containers have not yet been tested and evaluated for their use in collections. Because PET jars may come from the factory coated with dust of unknown composition, they should be thoroughly washed before use in the collection. Acrylic (e.g., Plexiglas) containers are unacceptable for long-term storage of specimens because they warp and crack, and because the preservative diffuses through the acrylic material (van Dam et al. 2000).

A comparison of plastic container materials is provided in Table 6.

Containers for Large Specimens

The traditional containers for large specimens in herpetological collections used to be large glass or ceramic crocks (Zweifel 1966). Although these are no longer in production, some are still in use in museums. They are difficult to seal properly, awkward to move, and are not an efficient use of space. In addition, the glaze on ceramic crocks eventually deteriorates from contact with the preservative, resulting in leakage (Smith 1965). Suzumoto (1992a) recommended using closed-cell foam weather strip tape or extruded polyethylene tape for replacement gaskets for large glass and ceramic containers.

The preferred containers for large herpetological specimens are stainless steel tanks (see *Appendix III*). The major drawbacks to these tanks are that (1) the

Table 6. Plastic containers.

Name	Identifying marks	Characteristics
High density polyethylene	HDPE	Resistant to fixatives and preservatives; susceptible to UV embrittlement; good resistance to oxygen permeance
Low density polyethylene	LDPE	Unsuitable for collection storage
Polyethylene terephthalate	PET, PETE	Resistant to fixatives and preservatives; relatively oxygen impermeable; lightweight; difficult to find good closures
Polypropylene	PE, P/E	Durable but flexible; very good for closures
Polystyrene	PS, P/S	Rigid, clear; permeable to water and oxygen; good for storage of dry materials but susceptible to damage from compounds containing benzene rings
Polyvinyl chloride	PVC	Embrittles; offgasses acids; unsuitable for use in collections

specimens cannot be seen without opening the tank; (2) tanks are hard to seal tightly; (3) the compressible gaskets fail after a few years from contact with the preservative; (4) many specimens must be handled to find the one you want; and (5) stainless steel tanks are expensive. Tanks should be housed on platforms with casters to they can be easily moved in the collection. Gaskets should be inspected regularly and replaced when they show signs of failure.

Tanks may be constructed from plywood and lined with epoxy or fiberglass (Dundee 1962), but these will not be as reliable as stainless steel tanks.

High-density polyethylene (HDPE) buckets or barrels are a low-cost alternative to steel tanks (see Table 6). Legler (1981) first proposed the use of plastic containers in herpetology collections; subsequently, spontaneous cracking of some of these containers was reported (Schueler and Aniskowicz 1982), but the cause of the cracking was not identified. Polyethylene becomes brittle when exposed to ultraviolet light. Containers that have been used outdoors for any length of time are not suitable for collection storage. Polyethylene containers should not be stored near windows, under fluorescent lights that lack ultraviolet (UV) filters, or near any other source of UV radiation. HDPE containers that have not been exposed to UV radiation and are protected from UV sources in storage are preservative resistant and will hold up reasonably well in collections, although HDPE does allow some permeance of oxygen that may affect preservative quality over time. Purchase containers with good seals (e.g., threaded lids with gaskets). See *Appendix III* for sources of HDPE containers. As with any large container in the collection, the condition of the buckets and barrels should be monitored regularly.

Carts and Trolleys

To move containers of fluid-preserved specimens safely, use a smooth-rolling cart or trolley. Carts and trolleys should be sturdy enough to hold the weight of a load of containers of specimens without bending; should have a lip or other restraining device to keep containers from falling off, be small enough to easily negotiate aisles and doorways in the collection storage area, and have large wheels (preferably with pneumatic tires) for smooth rolling. Clark et al. (1994) described a safety trolley for moving large containers of fluid-preserved specimens.

ALLOCATION OF SPECIMENS TO CONTAINERS

A variety of standard sizes of jars and other containers should be available for use in the collection. Using containers of standard sizes makes the process of replacement of containers and closures more cost efficient, and makes it possible to accurately plan collection expansion or relocation. Purchasing containers in large quantities directly from the manufacturer will reduce the cost per container. The standard container sizes recommended for herpetological collections are listed in Table 7.

The smallest standard size container that will reasonably hold the specimen should be selected. The container must be large enough that digits and tails of the specimens inside it are not stressed, but not so large that excessive amounts of preservative and shelf space are used. Select a container that will allow for a 7:3 ratio of fluid volume to specimen volume (Simmons 2014). Too

many specimens in a container will result in increased time to locate needed specimens, unnecessary handling of specimens, increased risk of damage to specimens, and significant dilution of the preservative (Taylor 1981a).

Table 7. Recommended standard containers for herpetological specimens.

<i>Container type</i>	<i>Closure type</i>	<i>Sizes</i>	<i>Use of container</i>
Glass jars	Polypropylene (P/P), continuous thread	8 ounce (0.5 pint) 16 ounce (pint) quart 1 and 2 liter 0.5, 1, 2, 3, and 5 gallon	Fluid preserved specimens
Glass jars	Polypropylene (P/P), continuous thread	40 x 73 mm 50 x 85 mm 58 x 66 mm	Cleared and stained specimens in glycerin
Glass vials, screw cap, continuous thread	Polypropylene (P/P) or PTFE, continuous thread	14 x 45 mm 17 x 60 mm 21 x 70 mm 25 x 95 mm 28 x 70 mm	Fluid preserved or cleared and stained specimens
Glass shell vials	Polyester fiber plug (submerged in larger glass container with P/P threaded closure)	15 x 45 mm 17 x 60 mm 19 x 65 mm 21 x 50 mm 21 x 70 mm 24 x 90 mm	Fluid preserved specimens
Stainless steel tanks	Compressible gasket; clamp lid	18 to 50 gallons	Fluid preserved specimens
Polyethylene bucks and barrels	Threaded lid with compressible gasket	5 to 50 gallons	Fluid preserved specimens
Polystyrene boxes	Polystyrene lids, hinged or not hinged	Variety, depending on specimen size	Skeletons, dry skins
PET boxes	Hinged PET lids	Variety, depending on specimen size	Skeletons, dry skins
Acid-free paper or board boxes	Acid-free paper or board	Variety, depending on specimen size	Skeletons, dry skins
Polyethylene bags	Heat sealed	Variety, depending on specimen size	Skeletons, dry skins

The container label should be pressed flat against the inside of clear containers, without overlapping itself or any part of a specimen, so that it can be easily read from the outside.

Fill the container to 90% of its volume for ethanol-based preservatives, and to 95% of its volume for water-based preservatives to reduce stress on the closure from temperature fluctuations. An increase in temperature increases the air pressure in the headspace of a sealed jar, which results in stress on the closure. The internal jar pressure depends on the vapor pressure of the fluid and the thermal expansion rates of the jar, the fluid, and the air above the fluid (Horie 1994; van Dam et al. 2000). For example, at 20°C, ethyl alcohol has an expansion coefficient forty times higher than glass, and water has an expansion coefficient eight times that of glass. For water or ethyl alcohol, an increase in temperature means a rise in the fluid level in the jar, which causes the compression of the air in the headspace, which puts stress on the closure and seal. The ratio of fluid volume to headspace is important. One reason to fill containers is to reduce the amount of air available for oxidation, but the higher the jar is filled with fluid, the greater the compression of the air in the headspace, and the greater the internal pressure.

When all containers are filled to a uniform level, those that lose fluid from evaporation or leakage are detected more readily. In the event that part of an exceptionally long specimen protrudes into the head space above the preservative, cover it with a layer of cheesecloth that has its edges below the level of the preservative, so that the specimen remains moist.

Some Tricks for Removing Jar Closures

Threaded Lids. When attempting to open a recalcitrant lid, grasp it so that your fingers wrap around the edge of the lid as completely as possible. This applies even pressure all around the edge. Wrap a wide rubber band around the lid to give a better gripping surface, or use an elastic jar opener. If this fails, try loosening the lid by running warm water over the jar, by tapping around the edge of the lid with a wooden stick, or by inverting the jar and striking the lid squarely on a flat surface.

Wire Bail Clamped Lids. Loosen the clamp, pull the bail off of the lid, and remove the lid. If the lid is stuck, twist it, tap around its edge, run warm water over the lid, or carefully insert a knife blade between the gasket and the lid to break the seal (the use of bottle forceps for this purpose is common but not officially sanctioned).

General Instructions for Handling Fluid-preserved Specimens

Specimens preserved in fluid, if well cared for, will probably keep indefinitely. Improper handling, especially desiccation, can quickly undo the work of centuries of preservation and care. When working with alcohol preserved specimens, frequently submerge them in a tray of the preservative (not water), or cover them with a cloth moistened with the preservative.

If a tail, limb or other appendage breaks off of a specimen, tie it to the venter with soft string (the type for stringing tags). Specimens with limbs broken but still attached should have the loose part tied to the body.

Specimens with openings in the body wall large enough for internal organs to come free should have the opening bound in string or sewn shut with a good quality thread.

Because the preservative surrounding the specimen is also part of the specimen (see above), change the preservative only when the solution presents a danger to the specimen (e.g., it becomes too acidic or too basic, or so discolored that it will stain the specimen). When changing old or contaminated preservative solutions, rinse the specimens in fresh preservative or in water (but do not soak them in water) before placing them in fresh alcohol.

When specimens are placed in containers, position them towards the bottom of the container so that if the fluid level drops from evaporation or a leak, the specimens will not be damaged until the loss of preservative is severe. Of course, before it gets to the severe point, the alert collection manager will take steps to ameliorate the problem.

Specimens should not interfere with the placement of the label in the container. When returning specimens to a container, put them in one at a time, keeping the label properly aligned. Gently shake the specimens down with forceps if necessary, watching for spaces that can form between specimens that might prevent some individuals from being nearer the bottom of the container than they could be.

General Instructions for Handling Larvae and Eggs

With few exceptions, larvae and eggs are kept in 10% buffered formalin (see *Fixation and Preservation*). Work with formaldehyde only in a well-ventilated workspace, preferably in front of a fume collector, and wear eye protection and formaldehyde resistant gloves (see *Formaldehyde Safety*). Keep specimens moist while working with them by keeping them in a tray of water or frequently submerging them in a formalin solution. Larvae and amphibian eggs are exceptionally fragile—handle them with a teaspoon or small tea strainer rather than forceps.

Keep larvae and eggs in shell vials with their catalog tags visible from the outside. Fill the vial with preservative and plug it with polyester fiber, then submerge it in a jar of preservative. Larvae should be stored head down or lying flat.

Procedures for Handling Specimens in Glycerin

Glycerin preparations are usually cleared and stained and are very fragile. Keep cleared and stained specimens in individual containers. Use external labels on the vials to avoid damaging the specimen with the label. Add a few crystals of thymol ($C_{10}H_{14}O$) or a few drops of camphor to each container of glycerin-preserved specimens to prevent bacterial or fungal growth. Thymol is a toxic chemical—use it only under a fume hood, do not inhale it, avoid skin contact with thymol dust or crystals, and wear eye protection when working with it.

ALLOCATION OF SPECIMENS IN THE COLLECTION

Specimens in standard size containers with proper labels are shelved in the collection storage area when not being used. Specimens (except types) are generally arranged alphabetically by family within each order, and within each family, alphabetically by genus and species. Within a species, specimens should be arranged by geographic locality. Specimens identified only to family are shelved after the last alphabetical unit in that family group. Although various “phylogenetic” arrangements are used in some collections, these are nonsensical, as a true phylogenetic arrangement would necessitate branching shelving (Simmons 2013). Use of an alphabetical system simplifies retrieval of specimens from the collection, thus enhancing collection use.

The traditional arrangements of collections date back to the time before electronic databases, when locating a specimen in a collection just by catalog number was time consuming. A good database should include a field for specimen location in the collection. As collections grow and storage areas become increasingly crowded, it is likely that traditional arrangements of the collection storage array will have to be abandoned in favor of arrays that make better use of space and arrange specimen containers for more efficient monitoring (Simmons 2013), using the database as the primary means of locating individual specimens.

Each taxon on the shelf may be delimited with a spacer made of wood, metal, or plastic to reduce errors caused when containers are accidentally shifted left or right. These spacers should be about a half-inch square and the same length as the depth of the shelf. Use of spacers makes the units in the collection more obvious, and reduces many common shelving errors. It is preferable not to split a taxon between two shelves unless the containers of the taxon occupy more than one shelf. Specimen containers should be spaced on the shelves to allow room for collection growth without requiring massive rearrangement of the jars.

Containers of formaldehyde-based preservatives should be housed separately from other specimens.

Holotypes and Paratypes. Types should be kept in a separate area of collection storage. Types are arranged alphabetically by genus and species without regard to family group. Type specimens are the most valuable material in the collection, and should be handled with special care. The specimen's type status is noted on the label by the word holotype or paratype. The scientific name on the holotype label is the original name for which type status is applicable, even if that name has been synonymized. Labels for paratypes may bear the current scientific name as long as the label is clearly marked “Paratype of <Original name>.”

Containers bearing holotypes are traditionally marked with a red ribbon tied around the neck of the jar or affixed to the outside of other types of containers. Containers bearing paratypes are similarly marked with blue ribbon.

Specific permission must be obtained from the curator before types may be removed from their containers for examination or sent out on loan.

Large Containers. Large containers (tanks, crocks, buckets, and barrels) should be numbered for identification. Keep a list of the catalog numbers of the specimens in each large container, organized sequentially. A small standard size jar with a dry label bearing the container number of the specimen should be housed with the regular collection to facilitate finding large container specimens. If possible, specimens should be allocated to large containers by some logical grouping (e.g., genus, family, or order). Nevertheless, due to the expense, most collections have a limited number of large containers and their contents end up fairly well mixed after a few decades of specimen allocation.

If specimens protrude above the level of the preservative, cover the protruding parts with a layer of cheesecloth or soft double-washed cotton cloth to wick preservative over the specimen. Nylon mesh bags may be used to group specimens together within large container. Do not overfill large containers with specimens—maintain the same 7:3 fluid volume to specimen volume that is maintained in small containers. Large containers are more susceptible to preservative loss from evaporation than are small containers, due to their increased surface to volume ratio and seal length. Check the fluid volume of large containers regularly.

Specimens in Glycerin. Specimens in vials and small jars of glycerin are allocated to shelving, cabinets, or drawers arranged in the same order as the regular collection. Stand the vials upright in cardboard tubes (Fig. 23) or with some other support to keep them upright (Ratcliffe and Messenger 1992).



Figure 23. Support system for vials.

Dry Skeletal Material. It is preferable to mark individual bones of skeletons with the specimen's catalog number to avoid mixing bones up between specimens. Use archival ink and an extra-fine tipped pen, and write the numbers on non-diagnostic parts of the bone. House skeletons in polystyrene or acid-free cardboard boxes with well-fitting lids. Arrange the boxes in trays or drawers in cabinets in the same order as the regular collection. Very small bones should be in small, lightweight vials made of polyethylene or polystyrene, or in gelatin capsules placed inside the boxes. Do not put glass vials in the containers with skeletons, as the movement of the vial will damage the bones. Stomach contents, skin, the hyoid apparatus, and other fluid preparations from skeletonized specimens should be housed in vials in standard jars in the regular collection, not with the dry skeletons. Monitor skeletal specimens regularly for signs of pest activity.

Skeletal Material in Fluid Preservative. Identify the preservative in the specimen record. Small or fragile specimens should be placed in a shell vial with a polyester fiber plug, inside a standard size jar of preservative.

MUSEUM COLLECTION MANAGEMENT

It requires a very unusual mind to undertake the analysis
of the obvious.

—Alfred North Whitehead (1925:4)

The basic functions of collection management are:

1. To permanently associate the individual specimens with their data.
2. To maintain the specimens and data records in optimum condition, preserving the maximum amount of information possible.
3. To make the specimens and data available to qualified users.

While collection managers are ephemeral beings, collections and museums are comparatively eternal. The entries made in the catalog today, or the note scribbled on a piece of paper and stuck in a jar explaining what happened to the contents must be interpreted after 50 or 100 or 200 years with the same veracity as the day they were written. Unfortunately, most of our mistakes become part of the collection, too.

Responsibility of the Collection Manager to the Collection

The primary responsibility of the collection manager is the care and maintenance of the collection and collection data. Neglect will result in loss of or irreparable damage to specimens and data, or separation of the specimens from their data. Even the smallest, most casual errors may be perpetuated for years. Within a short time of their occurrence, many errors are impossible to correct. The collection manager must:

- Keep procedures logical and standardized.
- Protect the integrity of the collection and the data.
- Make specimens and data available for use.
- Keep records of chemicals and procedures used to process and care for the collections.
- Monitor the storage environment.
- Keep records of collection activities.

- Strive to improve standards of collection management.
- Keep the collection and preparation laboratory organized and stocked with necessary supplies.

Responsibility of the Collection Manager to the Collection Users

A systematic collection is an irreplaceable scientific resource; properly documented specimens have intrinsic value, scientific value, and monetary value (Shelton and Simmons 2000). It is the responsibility of museums to manage and care for their collections as a public trust (Malaro and DeAngelis 2012).

It is presumptuous to think that we know all of the questions that can be answered by using preserved specimens and their data. In addition to the variety of current uses for specimens and data, new uses will undoubtedly be found in the future. The collection manager must keep the collections in a state that will enable them to provide maximum usefulness in the future. Collection use requiring destructive or consumptive sampling must be carefully evaluated.

LOANS, GIFTS, AND EXCHANGES

Loan Policy

The institution should have a clear and concise policy regarding what will be loaned, to whom, for how long, and for what purposes (see *Collection Management Policy*). Historically, most borrowers of scientific specimens were workers in other systematic collections, so loaning institutions could assume that borrowers knew how to care for specimens. Increasingly, people are using systematic collections who do not know how to handle specimens and who do not have the facilities to care for them. A concise sheet of loan conditions and instructions for handling specimens should be included with each outgoing loan (see Fig. 24).

Although there was long a *quid pro quo* relationship with regard to collection resources among museums, many collection users now offer little or nothing in return to the institution that is faced with the expense of maintaining the collection and making it available to the scientific community. Consider, for example, requests for tissue samples. The person requesting tissue samples likely will not have specimens to loan or exchange in return, and the use of tissues is destructive. In the future, museums will have to find ways to recover the costs of caring for specimens and making them available to those members of the community who do not contribute to the growth or maintenance of collections.

A loan policy should include the following concerns:

1. Specimens are loaned to institutions, not to individuals. Specimens on loan may not be transferred or removed from the institution to which they are on loan without written consent from the loaning institution.
2. Although loans are made to institutions, the individual who requested the loan is still responsible for the welfare of the specimens on loan. Students wishing to borrow specimens must have their academic supervisor co-sign their loan requests.

GENERAL LOAN INSTRUCTIONS AND CONDITIONS

1. Sign and return the marked copy of the loan invoice promptly.
2. The borrower agrees to protect and properly preserve material on loan.
3. Material must be returned by date state on invoice unless a renewal request is granted.
4. Material on loan is the responsibility of the institution named on the invoice.
5. Material on loan may not be transferred or transported to another institution without prior permission.
6. Tags and labels may not be removed without prior permission.
7. Any dissections must be approved in writing.
8. The loaning institution must be acknowledged in any publications or reports based on the borrowed material.
9. Send a copy of any publication or report resulting from use of this material to the loaning institution.

INSTRUCTIONS FOR HANDLING FLUID-PRESERVED SPECIMENS

1. Maintain specimens in the same preservative in which they are received (usually 70% ethyl alcohol or 10% neutral buffered formaldehyde).
2. Do not put specimens in denatured alcohol or any other type of alcohol without prior permission.
3. Do not submerge specimens in water.
4. Do not allow specimens to dehydrate.
5. Keep specimens away from heat.
6. Do not expose specimens to ultraviolet (UV) radiation.
7. Pack specimens carefully for return shipment.
8. Drain standing liquid from packages before sealing (do not ship specimens with a pourable amount of fluid in the package).

INSTRUCTIONS FOR HANDLING DRY SKELETAL SPECIMENS

1. Take care to keep all skeleton elements together.
2. Do not clean bones without prior permission.
3. Pad skeleton elements carefully for return shipment.

Figure 24. Loan conditions and specimen handling directions.

3. Specimens on loan may only be used for the purposes for which they were loaned, unless written permission is obtained.
4. Copies of loan invoices must be signed and returned promptly.
5. Written permission is required to make any modifications to a specimen, including opening the body cavity, removing anything from the body cavity, or preparing any parts. Any parts removed must be labeled and returned with the specimen.

6. Specimens must be properly packed for return to the loaning institution.
7. The entire loan should be returned at the same time, if possible. Large loans should be returned in more than one shipping container.
8. All taxonomic changes and re-identifications should be communicated to the loaning institution in writing. Copies of any publications or reports based on the borrowed specimens should be sent to the loaning institution.
9. Specimens should be cited in publications by catalog number, using the correct symbolic code (Leviton et al., 1985; see also *Appendix III*).
10. For a detailed discussion of conditions for outgoing loans, see Merritt (1992) and Simmons (2006).

Categories of Loans

There are four categories of loan transactions:

1. Outgoing loans.
2. Receipt of returned loans.
3. Receipt of specimens on loan for use by a member of the museum staff or a visitor.
4. Return to sender of loans received for use by a member of the museum staff or a visitor.

To this some museums add a fifth category—internal loans, for specimens used in the institution outside of the administrative unit responsible for the collection.

Loan shipments entering or leaving the United States must be reported to the US Fish and Wildlife Service by means of a 3-177 declaration (see *Appendix I*). For shipments of non-CITES listed cataloged specimens from one museum to another, the 3-177 declaration must be filed with the appropriate Fish and Wildlife office within 180 days. Most institutions with heavy loan traffic file the necessary 3-177 forms twice each year; other institutions prefer to file the forms as each shipment is sent or received. If the shipment contains CITES listed species, the CITES permits must be obtained for the shipment and the 3-177 declaration must be filed at the time the loan is sent or received. The US Fish and Wildlife Service webpage has a listing of regional offices, contact information, 3-177 forms, and instructions. The CITES webpage has a list of species covered under CITES and the CITES regulations. Both web pages are listed in *Appendix I*.

Loan transactions (for both the initiation and return of loans) should be numbered sequentially and invoiced for tracking and record keeping. The transaction invoice provides the legal documentation of the movement of the specimens. The museum copy of the invoice is archived as part of the museum's permanent records.

The following considerations should be evaluated for loan requests:

- ***Number and Size of Specimens Requested.*** Is the number of specimens reasonable? Does it include all of the specimens in the collection of a particular taxon? Are similar specimens available in other institutions? What will be the cost of retrieving, packing, and shipping the specimens? Some specimens (such as frogs) are relatively easy to pack and inexpensive to ship compared to others (for example, tortoises).

Some large requests may be better divided into two or more separate loans, with one installment being sent upon the return of the previous installment. Most museums will not loan out all of their specimens of a single species at the same time.

- ***Nature of the Specimens Requested.*** Are the requested specimens holotypes or paratypes? Are they specimens of a species poorly represented in the collection, or an extinct species? Is the specimen a voucher for an audio recording or image?
- ***Condition of the Specimens Requested.*** Are the specimens too old or fragile to be packed and shipped? Are they a preparation type that does not travel well?
- ***Use of the Specimens.*** Is the receiving institution equipped to properly care for the specimens requested? What does the borrower intend to do with the specimens? Any dissections, molding and casting, photographs for publication, or use in exhibition must be approved in advance.
- ***Project Design.*** Are the specimens requested really needed by the researcher? The project should be outlined in the letter of request. Is the borrower an overachiever asking for an unreasonable number of specimens, or someone proposing an unrealistic project?
- ***Loan Record of the Borrower.*** By keeping a borrower record of loan activity, you can evaluate the chances of seeing the loan returned within your lifetime. If a borrower already has a large number of specimens on loan, insist that they be returned before more specimens are sent. If specimens have been mishandled or abused by the borrower, or packed improperly for return, you should educate the borrower or deny future loan requests.
- ***Location of the Borrower.*** Particularly for large, hard to pack, or expensive to ship requests, it is sometimes preferable for the borrower to visit the museum rather than to send specimens on loan.
- ***Priority Use of Specimens.*** Has another researcher reserved the specimens for a current, on-going, reasonable project?

The Loan Invoice

Loan invoices should be printed on standard size, white, acid free 100% cotton rag paper. The original (signed) loan invoice is part of the historical record of the activities of the museum; once the loan is completed the invoice should be archived.

The loan invoice consists of four copies:

1. The first copy is maintained on file as a record of the transaction while the loan is active.
2. The second copy is mailed to the borrower separately from the specimens. The borrower checks the specimens against this invoice when the loan is received. This copy is signed and returned to the loaning museum to be archived as a permanent record of the loan transaction.
3. The third copy is retained by the borrower.
4. The fourth copy is used as a packing list and is sent with the specimens.

Do not use self-carbonized paper or colored paper for loan invoices, as these are not archivally stable. Loan invoices should be generated using a printer

that communicates with the collection database in order to produce multiple originals and minimize errors.

The loan invoice should contain the following information:

1. Name, address, postal code, telephone number, and email address of the lending institution.
2. Name, address, postal code, telephone number, and email address of the borrowing institution.
3. Name of person the loan is to be used by or who requested the loan.
4. Date the loan was sent to the borrower.
5. Means of shipment (e.g., US Mail, Federal Express, air freight, hand-carried).
6. Loan number.
7. Catalog number, scientific name, and locality data for each specimen in the loan.
8. Preparation type of each specimen and how it is to be housed (e.g., in 70% ethyl alcohol).
9. Date the loan is to be returned.
10. Space for borrower to sign upon receipt of the loan.

Procedures for the Preparation of Outgoing Loans

Specimens should be wrapped in a layer of cheesecloth (gauze) so that each specimen is covered. The cheesecloth-shrouded specimens are then placed in a polyethylene bag, and moistened with the appropriate preservative. Polyethylene for packing specimens can be ordered as individual bags or in long tubes that can be sealed with a heat sealer to make bags. If bags are to be tied shut, use two mil polyethylene. If a heat sealer is used, select a four mil polyethylene. The thicker polyethylene provides better protection for the specimens, but must be sealed with a heat sealer. Do not attempt to close plastic bags using rubber bands (which deteriorate from contact with preservative), paper clips or staples (which oxidize), or with string (which stretches when wet). Do not use zipper bags (e.g., Zip-Loc) or wire clamp bags (e.g., Whirl-Pac), as these do not seal well enough to withstand the stresses of shipment. See *Appendix III* for suppliers of plastic bags, polyethylene tubing, and heat sealers.

Each bag should be labeled and well-sealed. No excess preservative should remain in the bag. If a bag leaks, the preservative may damage the shipping container. Seal the bag of specimens inside a second plastic bag, and pack the specimens in a sturdy box for shipment, padded with packing material. Never pack a container so that a plastic bag of specimens is in direct contact with the outside wall of the container. Although specimens should be packed just prior to shipment and unpacked immediately upon receipt to avoid dehydration risks, well-packed specimens will survive several weeks when properly sealed in double plastic bags.

Use new, clean, unmarked boxes for shipping specimens—reuse of old boxes may result in the loss or misdirection of a shipment, and storing old boxes in the lab may introduce pests into the collection. The preparation laboratory should have standard sizes of new boxes on hand for shipping (see *Appendix III* for recommended suppliers).

1. *Select the specimens to be loaned.* Loan requests must be approved by the appropriate authority (usually the curator). Be sure that the person approving the loan is aware of any paratypes, holotypes, unique specimens, voucher specimens, or exceptionally fragile specimens included in the request. Once the request is approved, retrieve the specimens from the collection storage area, and bring them to the preparation laboratory.
2. *Complete the loan form.* Have the complete copy of the loan form in front of you to check off the specimen's catalog numbers one-by-one as they are packed.
3. *Wrap each specimen in at least one layer of cheesecloth.* Several specimens may be rolled up in one length of cheesecloth. Keeping in mind the size of the plastic bags and packing containers available, spread out a layer of cheesecloth on a flat surface, then lay out the specimens so that fingers, toes, and tails will not be stressed (Fig. 25). Fold the edges of the cheesecloth over the specimens and roll the package up securely but not tightly. It may be necessary to pad sharp, protruding claws (e.g., turtles, some lizards) or spines (e.g., *Phrynosoma*; see Montanucci 1994). This can be done by wrapping the sharp parts with cheesecloth, or by padding them with polyester fiber and then wrapping with cheesecloth.
4. *Insert one or two rolls of wrapped specimens into a polyethylene bag.* Do not force specimens into a bag that is too small.
5. *Pour a small amount of the appropriate preservative solution* into the bag with the specimens, and thoroughly moisten the cheesecloth. Carefully decant the fluid, so that no standing fluid remains in the bag.
6. *Seal the bag.* Use a heat sealer with a 1/8 to 1/4-inch seal width.
7. *Place the sealed bag inside another bag* along with some absorbent material (e.g., a piece of absorbent padding), insert an address label between the bags so that it is visible from the outside, and seal the second bag (Fig. 26). Depending on the thickness of the plastic and the size and weight of the specimens, a third bag may be appropriate as well.
8. *Larvae* should remain in fluid at all times to avoid dehydration. They should be packed in screw-top vials with good closures. An alternative is to use a loose packing of clean polyester fiber, moistened with the preservative above, below, and around the larvae inside the vial, taking care not to compress the specimen. Include a paper label bearing the specimen lot number inside the vial. After screwing the lid on tightly, tape the vial/lid junction to prevent it from coming loose. Seal the vial inside two plastic bags with an address label.
9. *Dry skeletons* should be packed in a sturdy acid-free cardboard or polystyrene box with clean polyester fiber padding. Small bones should be placed in gelatin capsules so they do not become lost in the packing material. Tape the lid to the box so that it does not come open during shipment.
10. *Cleared and stained* specimens in glycerin should be shipped in a small vial of glycerin. Tape the lid to the vial and seal the vial inside two plastic bags with an address label visible from the outside.
11. *Pack the plastic bags of specimens in a sturdy cardboard box.* Do not allow any of the specimen packages to be in direct contact with the outer wall of the container, as this would place them at greater risk if the container was damaged in shipment. Use a packing material that will maintain its bulk during shipment. I recommend the use of plastic packing pellets. Do not use biodegradable packing pellets—they are

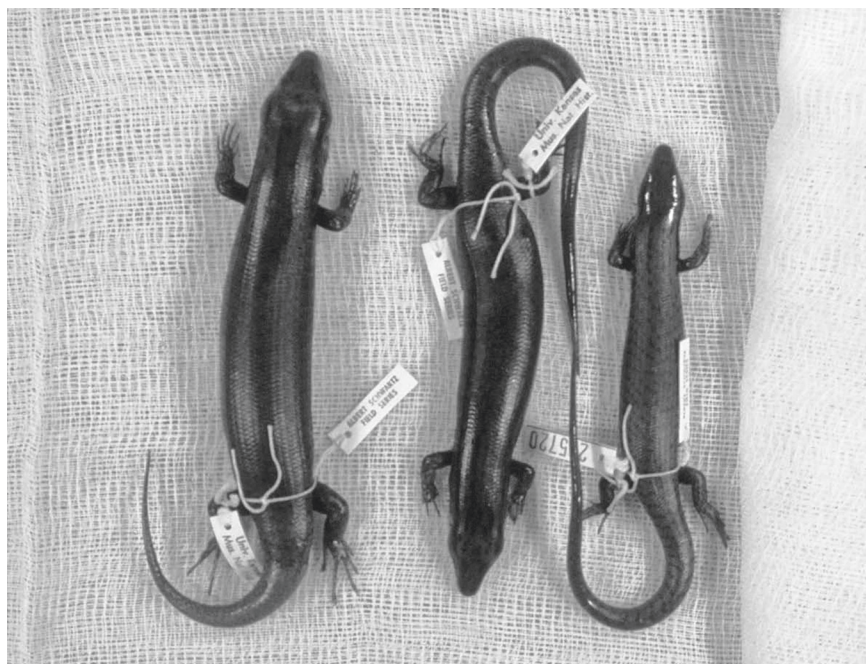


Figure 25. Positioning specimens for packing.



Figure 26. Packed specimens with address label.

made of a cornstarch-based material that attracts pests and dissolves upon contact with preservative fluids or water.

12. *Enclose a return address label* and shipping copy of the loan invoice inside the package.
13. If shipping CITES listed specimens, attach a copy of the institutional CITES permit to the outside of the package in an envelope labeled "CITES permit."
14. If shipping the package outside of the United States, complete the required customs declaration forms and attach them to the package. Describe the contents as "Preserved scientific specimens" and list the value as "No commercial value." If a commercial value is listed, the recipient may have to pay duty to retrieve the shipment from customs.
15. *Seal the container* with a sturdy plastic box sealing tape. Do not use paper tape (which has humidity sensitive adhesives), masking tape (which has weak adhesives), or strapping tape (which rapidly deteriorates with age and exposure to UV).

Shipping Fluid-preserved Specimens

As of January 2011, the International Air Transport Association (IATA) regulations include a special provision for shipping non-infectious natural history specimens (Bentley 2011). To qualify, the specimens must be wrapped in cheesecloth or paper towels that are moistened with ethyl alcohol, isopropyl alcohol, or 10% formalin, and be placed in heat-sealed plastic bags containing no more than 30 ml of free liquid per bag. The bag of specimens must be heat-sealed inside a second bag containing absorbent material, and the double-bagged specimens must then be packed in a sturdy box with cushioning material. The total quantity of flammable liquid per shipping container may not exceed 1 liter. The shipping containers must be marked as "scientific research specimens, not restricted, Special Provision A180 applies." The words "not restricted" and the number "A180" must also be included in the description of the substance on the air waybill (air consignment note) when an air waybill is issued (Bentley 2011). The special IATA provision does not include checked or carry-on airline baggage. Appropriately packed and labeled packages may be sent by USPS, FedEx, and DHL, however, some international restrictions may still apply, such as the need for mandatory veterinary inspections when shipping to certain countries (Bentley 2011).

Marking Containers of Specimens on Loan

Specimen containers and labels should never be sent out with loaned specimens. Specimen containers should be marked (see below) to indicate that the specimen is on loan, and promptly returned to the collection to serve as placeholders for the borrowed specimens.

Containers from which some or all of the specimens have been removed for a loan should be marked with a small loan slip attached to the outside of the container. The loan slip should contain the following information:

- The catalog number(s) of the specimen(s) removed.
- The loan number.
- The last name of the borrower.
- The date of the loan.
- The name or initials of the person who prepared the loan.

A sample format for a loan slip is shown in Fig. 27.

Returning Loans to Senders

When specimens are returned to the loaning institution, a return-of-loan invoice should be prepared to establish a paper trail documenting the return of the specimens. The sequentially numbered invoice is similar to the invoice used for outgoing loans. Include the original loan number on the document to aid the loaning institution in processing the return. The return-of-loan invoice should contain the following information:

1. Name, address, postal code, telephone number, and email address of the institution returning the loan.
2. Name, address, postal code, telephone number, and email address of the institution the loan is being returned to.
3. Date the loan is returned to the lender.
4. Means of transport of the shipment.
5. Return-of-loan number.
6. Original loan number from the loaning institution.
7. Catalog number and scientific name for each specimen being returned.
8. Provide a space for a representative of the loaning institution to sign upon receipt of the returned specimens to verify the completion of the transaction.

INTERNAL USE OF SPECIMENS

When specimens from the collection are used in-house by curators, staff, students, or visitors, they are usually removed from the collection for days to months at a time. Users should fill out an in-house specimen use form and affix it to the specimen storage container. If the entire container of specimens is removed from the collection room, place the in-house use form in an empty container of the same size and shelve it in the collection to mark the spot where the specimens should reside. This will aid in tracking specimens that are being used, remind the collection manager to retrieve specimens from users in a timely fashion, and in the case where an entire container is

SPECIMEN(S) WITHDRAWN FROM THE COLLECTION Division of Herpetology, Natural History Museum	
Numbers: _____ _____	
Loan Number: _____	Date: _____
Skin, skull, skeleton, alcoholic, larvae	
Withdrawn by: _____	

Figure 27. Container marker for specimens on loan.

removed, serve as a place-holder on the shelf. The in-house use form should be marked with the borrowed and returned dates and name of the borrower (see Fig. 28 for a suggested format). The completed in-house use forms are returned to the collection manager upon return of the specimens; the forms provide an index of in-house collection use and highlight areas of the collection that may need special evaluation of preservative concentration and container seals.

MIXING PRESERVATIVES

Preservative fluids should be mixed under a fume hood, in front of a bench-top fume collector, or in another well-ventilated area to prevent inhalation of the preservative vapors. Ground the container and the pump before dispensing bulk preservative (see *Fire Code and Safety Regulations*).

Use distilled or deionized water to mix preservatives—tap water contains minerals and other chemicals that will contaminate preservative solutions. Measure fluids in a clean, graduated container (do not attempt to measure

WITHDRAWAL FORM Taxon: _____ Number(s): _____ _____ Political unit: _____ Taken by: _____ Temporary location of specimen(s): _____ Date withdrawn: _____ Returned by: _____ Date returned: _____

Figure 28. Form for internal use of specimens.

fluids by reading the giant meniscus in a 5-gallon carboy sitting on the counter). Allow newly mixed fluids to sit for at least 24 hr after mixing before they are used. During this time the solution will reach equilibrium and return to room temperature (fluid temperature increases due to heat generated by mixing the preservative with water). Do not attempt to speed the mixing of preservative solutions by bubbling air through the liquid, as this may cause oxidation of the preservative.

Check the strength of alcohol solutions with a digital density meter or hydrometer (see Simmons 2014 for a discussion of determining fluid concentration). Digital density meters are expensive (see Appendix III), but are more accurate and much easier to use than hydrometers, and can be used on much smaller volumes of fluid. If neither a digital density meter nor a hydrometer is available, you can make an alcohol tester as described by Moore (1983, 1994) using plastic beads or heads of map pins that float in alcohol.

A hydrometer consists of a sealed glass tube containing a scale and a weight at one end. The tube is floated upright in the fluid to be tested (hence the volume of fluid must be sufficient to float the hydrometer), and the scale is read at the meniscus. Because fluid density changes with temperature, a correction must be calculated based on fluid temperature to determine alcohol concentration. To obtain an accurate reading, the hydrometer must be clean and dry before it is inserted into the fluid to be tested, and must be at the same temperature as the fluid (e.g., don't bring containers of specimens from a cool storage area into a warmer laboratory and immediately insert the hydrometer—allow time for the fluid and the hydrometer to reach the same temperature). The hydrometer must float free in the fluid, so it may be necessary to remove some or all of the specimens from the container of fluid to be tested. Place the hydrometer in the fluid, allow any air bubbles to dissipate, allow the hydrometer to float free, and read the scale at the point where the hydrometer intersects with the surface of the fluid (you must be at eye level with the meniscus to obtain an accurate reading). Refer to Waller and Strang (1996) for a chart showing density measurements, temperature, and volume percent of ethyl alcohol.

An alternative method for monitoring alcohol concentration is the use of a commercial product called the Alcomon Indicator System (see *Appendix III*). The system consists of two small polypropylene disks containing iron filings. Both disks sink if the concentration of ethyl alcohol is 60% or above; one disk sinks and one floats at concentrations of 50–60%; both disks float at concentrations below 50%. The disks can be left in the specimen containers for years, allowing a quick visual inspection to detect changes in alcohol concentration. A pair of Alcomon disks can also be used in a small glass tube for checking alcohol concentration (pers. comm. A. Bentley).

TOPPING UP CONTAINERS AND PRESERVATIVE CONCENTRATION

Because alcohol evaporates from preservative mixtures faster than water (Simmons 2014), as the fluid level drops in a container due to evaporation, the concentration of alcohol will decline rapidly. Containers should be topped up with an appropriate concentration of preservative to achieve the desired storage strength because continued topping up with stock solution will result

in an increasingly lower than desired concentration of preservative. Sendall and Hughes (1996) have provided a formula for use in correcting alcohol concentrations when topping up straight-sided containers. The formula is:

$$z(x + y) = ax + by$$

where z is the desired concentration of preservative (percent); x is the measured height of fluid in the container; a is the measured concentration of fluid (percent); b is the concentration of the solution being added (percent); and y is the amount of fluid to add (relative to container height). The equation is solved for y :

$$y = (z - a)x / (b - z)$$

For example, assume that a jar that contains 11 cm of ethyl alcohol (x) that has a concentration of 59% (a), determined by the use of a digital density meter. To top up the container and restore the fluid to a 70% concentration (z) using 95% ethyl alcohol (b), solve the equation for y , which will reveal that 4.84 cm of 95% ethyl alcohol (b) must be added.

$$y = (70\% - 59\%) 11 \text{ cm} / (95\% - 70\%)$$

$$y = 4.84 \text{ cm of } 95\% \text{ ETOH}$$

Containers should be filled to a standard height so that those losing alcohol can be immediately detected by visual monitoring (see *Allocation of Specimens to Containers*). When containers are determined to have lost alcohol, the fluid level should be marked with a grease pencil (china marker), which makes a durable but non-permanent mark on glass, before the container is re-filled. With a regular monitoring schedule, faulty containers and closures can be readily detected and replaced.

It can be very time consuming to check the alcohol concentration in each jar in a large collection. As a general rule of thumb, if the fluid level has declined by 10% or less from the standard fill level, the container may be topped up with stock solution, marked with a grease pencil, and monitored. If the fluid loss is greater than 10% below standard fill level, the concentration should be checked and the fluid brought back to the standard concentration by the method described above (see discussion in Simmons 2014). For this procedure to be effective, regular visual inspections of fluid levels must be made.

Notton (2010) proposed another method for managing fluid replacement in a large collection using standard sizes of containers, regular inspections, and a digital density meter to develop a topping-up table for replacing lost fluid.

REHYDRATION OF SPECIMENS

A variety of rehydration formulas and techniques have been published (see Simmons 2014 for a review). Some of these are successful for certain types of organisms, but after years of unhappy experiences, I have concluded that as a general rule, once a fluid-preserved specimen has become severely dehydrated, it should probably be maintained in that state unless it is

absolutely necessary to rehydrate it in order to use it. While dehydration is very damaging to fluid-preserved specimens; rehydration causes even more tissue damage, and can shorten the useful life of a specimen. In general, the softer a dehydrated specimen is, the more successful rehydration is likely to be. Specimens that are so dry they have become crisp and hard are far less likely to be successfully rehydrated. Several rehydration techniques are listed in Table 8—note that most of these employ a surfactant or other chemical to soften the tissues, which damages specimens. It is best to experiment on a junk specimen before attempting to rehydrate a valuable specimen.

By far the most successful technique I have found for rehydration of amphibian and reptile specimens is a slow rehydration process that exposes the specimen to a humid environment to soften it for several days before rehydrating it in clean water and returning it to the preservative (Simmons 2014; Singer 2014). Select a clean container with a good closure; add an inch or so of deionized or distilled water and a few crystals of thymol or a few drops of camphor to the water to prevent mold and bacterial growth. Place the dehydrated specimen inside the container on an open rack over the water (but not in contact with it) and close the container tightly. Allow the specimen to soften in the container for several days, monitoring the water and specimen for any indication of mold or bacterial growth. Once the specimen has absorbed enough water vapor to become soft, place it in a clean solution of deionized or distilled water for 24 hr, then stage it up by 20% steps to the desired strength preservative (following the protocol outlined in *Transfer of Specimens from Fixative to Preservative*).

UPDATING OR MODIFYING SPECIMEN RECORDS

Post-cataloging specimen preparation, correction or addition of locality or other collecting information, re-identification, synonymy of previous names, and the naming of new taxa are the principal reasons for needing to make changes to specimen records. When to make changes is often a judgment call for the curatorial staff. Generally, it is the responsibility of the curator, in consultation with the collection manager, to make these decisions. Factors to consider include:

- Nature of the specimen preparation.
- Relevance of new or corrected information.
- Reliability of re-identification or new name.
- Likely acceptance of a synonymy or a new name by the scientific community.
- Publication date of the new name (do not use a manuscript name on a label or in a database, as the name may be circulated accidentally before it is published by the author).
- The opinion of the curator as an expert systematist.

Collection workers and visitors who wish to recommend a change in specimen name or information should fill out a specimen record change form (Fig. 29) and give it to the collection manager. The collection manager then consults with the curator (if necessary) before making the change.

When making nomenclatural updates, update all specimen records. These records may include the database, container labels, and labeling of images

Table 8. Rehydration techniques.

<i>Technique</i>	<i>Reference</i>	<i>Comments</i>
30% ammonia and warm water	Traditional technique	Poor results; ammonia damages proteinaceous tissues
2:1 hydrogen peroxide and water	Traditional technique	Poor results; hydrogen peroxide damages tissues
3:1 isopropyl alcohol and water	Traditional technique	May initially soften tissues but results in further dehydration of tissues over time
Surfactant in aqueous solution	Banks and Williams 1972	Generally poor results on badly dehydrated specimens; surfactant chemicals difficult to rinse out of tissues
0.25% to 0.50% trisodium phosphate in water	van Cleave and Ross 1947	Usually unsatisfactory; trisodium phosphate damages tissues
Stage through 10%, 5%, and 0.5% acetic acid, followed by trisodium phosphate solution	Vogt 1991	Usually successful, but chemicals will damage tissues
50% polypropylene glycol or ethylene glycol	Marhue 1983	Unknown effects on tissues
10% solution of enzymatic drain cleaner (e.g., Drano Liquid Plumber) for 3 wks	Vogt 1998	Extremely damaging to tissues
Soak in Decon 90 or TEEPOL at 40°C; inject with preservative; remove air bubbles with vacuum pump	Waterhouse and Graner 2009	Fair to good results
Exposure to vapor from water treated with thymol until soft; staged by 20% steps from 100% deionized or distilled water to desired preservative strength	Singer 2014	Slow technique; requires monitoring for mold or bacteria; satisfactory results in most cases; recommended technique

SPECIMEN DATA CHANGE FORM	
Taxon:	_____
Number(s):	_____
Change identification to:	_____

Change status to:	_____
Change locality to:	_____

Other:	_____
NOTES:	
Name of researcher:	_____
Date:	_____
Approved by:	_____
Date:	_____

Figure 29. Form for changes to specimen data.

or recordings. The nomenclatural change may be recorded in the permanent handwritten catalog in pencil.

SPECIMENS EXCHANGED, GIVEN TO OTHER INSTITUTIONS, DESTROYED, OR LOST

The catalog and database should be annotated to record any specimen that is exchanged, given to another institution, destroyed, or lost. However, catalog numbers should never be re-assigned. Even when a specimen is destroyed, the catalog record of the specimen should be retained. The record that the specimen was once part of the collection is valid information, even if the specimen is no longer extant. When specimens are exchanged or given to another institution, record the institution's symbolic code (Levinton et al. 1985; see also *Appendix III*) and the specimen's new catalog number in the hand-written catalog and database. When a specimen is destroyed or lost, briefly describe what happened so that when a researcher in 50 or 100 years comes looking for the specimen, its fate can be determined.

GUIDELINES FOR COLLECTION GROWTH

Peale even preserved an unusual fish without a head, deeming it worthwhile to preserve that much until more could be learned.
—C.C. Sellers (1980)

The philosophy of collection growth was once quite simple—more was better. Any specimen from most anywhere was fair game for accession. If for no other reason, the rising cost of collection storage and information management for large collections have made this attitude obsolete, but ethical and legal considerations are also involved in collection growth. See Cato (1986); Fritts (1976); Malaro and DeAngelis (2012); and Simmons (2006) for discussions of museum acquisition policies.

The following criteria should be considered when presented with the opportunity to accession new material:

1. *Collection documentation.* Do the specimens have adequate supporting documentation (e.g., field notes, images, recordings)? Is the collector reliable and trustworthy?
2. *Specimen preservation.* Were the specimens adequately prepared in the field? Can you reasonably expect them to be useful scientific specimens in 50, 100, or 200 years in the future? Are the specimens worth the investment of time and materials necessary to process them and house them properly?
3. *Legal issues.* Does the donor provide appropriate legal documentation in the form of collecting, export, and import permits? If the specimens were collected outside the United States, does the documentation include a copy of the 3-177 declaration? The burden of proof that the specimens are legal lies with the institution receiving them (see Malaro and DeAngelis 2012 and Tompkins et al. 2010 for further discussion).
4. *Ethical considerations.* Were the specimens obtained in an ethical manner? Do they represent a last chance to preserve the biodiversity of a threatened ecosystem, or does their collection lend encouragement to the destruction of wildlife of a region? Is the addition of these specimens justifiable in terms of their potential scientific value?

5. *Need for the specimens.* Will this accession complement specimens already in the collection, or fill gaps in the collection? If the specimens would be of little use to the regular users of the collection, might they be better deposited in another institution? Are there other institutions that might benefit more from receiving the specimens? Is it important to accept the specimens as voucher specimens (Lee et al. 1982)? Merritt and Lidgard (2000) proposed a risk management model for acquisition and accession.

Guidelines for Exchanges

There are two types of exchange agreements. A one-time exchange is worked out with specific numbers of specimens or specific specimens in mind. An open exchange is an ongoing process. Some museums use open exchanges of paratypes in order to avoid having type material housed in only one collection, in case of disaster. As a general rule, open exchanges are discouraged because they can lead to inequality of expectations.

The following considerations must be taken into account when entering into exchange agreements:

- The exchange is not completed until both parties have the specimens in question in their possession.
- The documentation for the exchange should spell out clearly the conditions, terms, and time limits on the exchange agreement.
- Documents relating to the exchange should be signed by representatives of both institutions and archived as part of the permanent collection documentation.
- Copies of field notes and other specimen data should accompany the specimen in an exchange.

EVALUATION OF COLLECTIONS MANAGEMENT

Good collection management depends on regular evaluation of the collection management program. Annual statistics should be compiled to evaluate collection growth and use, particularly to identify underdeveloped or underused parts of the collection.

Evaluation of the Collection Management Operation

The effectiveness of the collection management program should be evaluated annually (Simmons 1986b). The following aspects of the operation should be considered:

1. Does the preparation laboratory provide a safe and secure environment for working with fluid-preserved specimens?
2. Does the collection storage area provide a safe, secure, and stable environment for the long-term care of the collection?
3. Do cataloging, processing, and allocation of specimens to the collection keep pace with incoming accessions?
4. Are specimens returned to a safe and secure storage environment immediately after use?
5. Are new accessions appropriately documented?
6. Is collection documentation stored in an archival, retrievable manner?
7. What is the response time for loan requests? It should be no longer than two to three weeks.

8. What is the response time for requests for specimen data?
9. Are there requests for specimen data that the data retrieval system cannot fulfill?
10. Has the growth of the collection since the last evaluation met expectations?
11. Do the collections care workers follow the standard operating procedures (SOP) that are in place?
12. Do the collection care personnel stay busy; do they have so much to do that the quality of the collection care provided is suffering?
13. Does the scientific community know that the collection exists? Evaluate user records to see if some parts of the collection are being overlooked and if so, take steps to correct oversights.

Evaluation of collection management and the state of the collection can be accomplished using the Collection Health Index or CHI (McGinley 1993; Williams et al. 1996). The CHI provides a way of rating the overall health of the collection by evaluation of storage units (in the case of fluid-preserved collections, the units are usually by shelf) rather than individual specimen assessments. It provides a management tool that is useful for assessing, describing, and comparing collection needs across a wide variety of collections within an institution.

Price and Fitzgerald (1996) have proposed a system of five categories that reflect the value of specimens based on scientific, cultural, and monetary considerations. Their classification system can be used to direct resource allocation within a collection and to justify the collection to administrators.

Annual Reports of Collection Management Activities

At the end of each calendar or fiscal year, prepare a summary of the previous year's collection management activities. Even if this is not required by the museum's administration, it is a useful way to analyze the collection management program and improve it. The information in an annual report should include:

- Number of accessions and total number of specimens accessioned.
- Number of specimens cataloged.
- Number of outgoing loans and number of specimens loaned.
- Number of loans and specimens returned.
- Number of loans and specimens received for staff, student, or visitor use.
- Number of loans and specimens returned to other institutions.
- Number of visitors on collection business and total number of visitor days.
- Number of inquiries received from the public (e.g., snake identifications).
- Number of requests for non-collection herpetological information (e.g., requests for data from bibliographic resources).
- Number of requests for collection data and total number of records provided.
- Number of published citations of specimens in the collection.
- Major collection management activities and improvements in the collection management operations.

To aid in tracking day-to-day collection management activities as well as compiling the information for reports, I recommend keeping a running list of activities by category on a clipboard in the preparation laboratory. Having

an up-to-date hard copy of collections management activities both speeds up annual reporting and provides a very handy reference for quickly answering queries concerning collection activities.

Track the following collections activities:

- ***Loans sent out (initiated).*** Include the loan number, name of the individual and borrowing institution, brief description of the loan contents (e.g., 12 snakes), and initials of the individual who prepared the loan.
- ***Loans returned (received).*** Include the loan number, the borrower's return-of-loan number (if any), date return was received, and initials of the person unpacking and reinstalling the specimens.
- ***Loans received for staff, student, or visitor use.*** Include the lending institution's loan number, name of lending institution, name of the staff member, student, or visitor who needed the specimens, date of receipt, and initials of person who unpacked the loan.
- ***Loans for staff, student, or visitor use returned to lending institution.*** Include the return-of-loan number, the lending institution's loan number, name of lending institution, date of return of specimens, and the initials of the person who packed the specimens for return.
- ***Receipt of gifts and exchanges.*** Include the date of receipt, name of sender, sender's institutional affiliation, brief description of specimens in the shipment, and initials of the person unpacking the shipment.
- ***Requests for collection data.*** For data requests that come directly to the collections manager, include the date that the requested data was sent, the method of data transfer (e.g., email, hard copy), general description of data, number of specimen records included, and initials of the person responding to the data request. Include annual summaries of data requests serviced by secondary data providers such as VertNet.
- ***Requests for non-collection information.*** Include the date the requested information was sent, method of information transfer (e.g., email, hard copy), general description of information (e.g., photocopy of a page of field notes), and initials of the person responding to the information request.
- ***Inquiries from the public, tours, etc.*** Include the date, number of members of the public served, and initials of the person dealing with the inquiry or tour.
- ***Visitors.*** Include the date(s) of visit, purpose of visit (for visitors on collection related business), and institutional affiliation of each visitor.

The nature of a collection management program is progressive and dynamic. Frequent evaluation will help assure that your program is able to respond to the changing demands from the scientific community, while still protecting the collection for future use.

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APPENDIX I. SOURCES OF INFORMATION FOR PERMITS

The Endangered Species Act (ESA) legislation is in 16 USC 1531 et seq., which is available on the internet through the US Fish and Wildlife Service (USFWS) website (USC is an abbreviation for United States Code, also called the Code of Laws of the United States):

www.fws.gov

The regulations for filing 3-177 declarations is in 50 CFR Part 14, which is available through the USFWS website (CFR is an abbreviation for Code of Federal Regulations). More information on importing wildlife and possible inspection fees may be found at:

<http://www.fws.gov/le/businesses.html>.

The Convention on International Trade in Endangered Species (CITES) legislation and regulations is in 50 CFR Part 23 (CITES), in the Federal Register (volume 72, number 163), and on the CITES website:

www.cites.org

Where to Obtain Information on What Permits are Needed

To get general information about permits and permitting processes, you may wish to contact individuals who have recently obtained permits for the area where you wish to collect by sending queries to PERMIT-L or the listservs run by the Society for the Preservation of Natural History Collections (SPNHC) or the Natural Science Collection Alliance (NSCA). However, keep in mind that laws and regulations governing wildlife change frequently, and do not rely just on the experiences of others. It is always best to confirm what permits are needed before initiating field work by contacting the appropriate authorities in the area where you wish to work.

Permit-L website:

gold.sdsmt.edu/mailman/listinfo/permit-l

SPNHC website:

www.spnhc.org

NSCA website:

www.nscalliance.org

Permits for US States and Territories

Each state and territory of the United States has its own regulations governing the collecting of wildlife for scientific purposes. A valid hunting or fishing license for the state or territory may be required in addition to a scientific collecting permit. Specific permits will probably be required for collecting in any protected area. Before initiating work in any US state or territory, check permit requirements specific to the state or territory—most of the requirements can be found on the internet by searching for state and territorial government websites.

Permits for Countries Other than the United States

It is strongly recommended that anyone wishing to collect in a foreign country work closely with a colleague in that country for help in obtaining permits and understanding applicable regulations. Contact information for permitting agencies in other countries can sometimes be provided by the US Embassy in that country (but it is important to note that the US embassy cannot assist in obtaining permits). A list of addresses and telephone numbers for US embassies is available on the US Department of State website:

www.state.gov/countries/

National Park Service Research and Collection Permits

science.nature.nps.gov/research

United States Fish and Wildlife Service Resources

Home page of the United States Fish and Wildlife Service

www.fws.gov

US Fish and Wildlife Service Permits page

www.fws.gov/permits/

US Fish and Wildlife Service lists of protected species

www.fws.gov/permits/SpeciesLists/SpeciesLists.html

Designated Port Requirement (United States Fish and Wildlife Service)

A current list of Designated Ports and the contact information for each port can be found on the US Fish and Wildlife Service website at:

www.fws.gov/le/designated-ports.html

US Fish and Wildlife Service Designated Ports

Anchorage, Alaska
Atlanta, Georgia
Baltimore, Maryland
Boston, Massachusetts
Chicago, Illinois
Dallas/Fort Worth, Texas
Honolulu, Hawaii
Houston, Texas
Los Angeles, California
Louisville, Kentucky
Memphis, Tennessee
Miami, Florida
New Orleans, Louisiana
New York, New York
Newark, New Jersey
Portland, Oregon
San Francisco, California
Seattle, Washington

Designated Port Exemption Permits

Specimens for scientific research can generally be imported or exported through a non-designated port if permission is obtained in advance. Applications for Designated Port Exemption Permits must be made using Form 3-200-2 from the United States Fish and Wildlife Service. For more information on Designated Port Exemption Permits and procedures for obtaining them, refer to:

www.fws.gov/le/designated-port-exception-permit.html.

Useful References Related to Collecting, Export, and Import Permits

Duellman, W. E. 1999. Perils of permits: procedures and pitfalls. *Herpetological Review* 30(1): 12–16.

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Convention on International Trade in Endangered Species (CITES)

CITES Home page

www.cites.org

CITES listed species

<http://www.cites.org/eng/disc/species.php>

APPENDIX II. FIELD WORK LISTS

This appendix is based on *Field Work Lists: Everything You Will Need for Camp or Trail* by John E. Simmons and Mark Robbins.

INTRODUCTION

The only thing worse than arriving at your collecting site and discovering that you do not have something you need is returning home and finding that you have been lugging around something that you didn't need. To help avoid these problems, the following lists may be used as checklists when preparing for a field trip. Obviously not everyone needs all of the things on the lists—the lists are to be used as guidelines and reminders for planning. Whenever possible, check with a recent traveler to the area where you are going to be sure that what you need can be purchased at your destination. If there are items critical to your trip that you are unsure of, take them with you.

Hints for Happy Camping

- Never waste space—small bundles use space more efficiently than large bundles.
- Pack small things inside of larger things.
- Remove most packaging from packaged goods.
- When padding around a liquid nitrogen tank, use only things that can be frozen and thawed safely (e.g., use towels, not a plastic drop cloth).
- When padding around containers of liquids, use only things that will not be damaged by leaks.
- Keep a detailed list of everything you pack in each bag or box. This will help you locate items easily and will be useful if you need to file a missing baggage claim or insurance claim.
- Take extra baggage labels with you (baggage labels are easily lost while traveling and may need to be replaced).
- Use your work address and telephone number on all baggage, never use your home address.
- Be sure to follow airline rules for weight and dimension restrictions on luggage, especially for carry-on luggage, and remember that the restrictions vary from airline to airline.

Money and How to Spend It

Traveler's cheques used to be the safest way to transport money, but their use has declined sharply in favor of cash, credit cards, and debit cards. If traveling outside of your home country, check with your card provider to confirm that you will be able to use your credit or debit card where you are traveling, and notify the provider of your travel dates. Do not take cashier's checks or international money orders when traveling overseas, as you will spend hours of valuable time trying to exchange them for local currency, and often will not be able to.

Always carry your funds in a money belt or money pouch securely on your person. Never leave cash or credit cards in a hotel room, in a pack or shoulder bag, or in a suitcase. Never carry money, credit cards, or anything else of value in a wallet in a hip pocket, in a purse, or in a fanny pack. Use a money belt or hidden pouch for all money and credit cards. Do not carry all of your

funds in the same place on your person. Spread funds among the members of your party, or use more than one money belt or pouch.

Before you depart for the field, empty your wallet of everything you will not need (e.g., library card, extra credit cards).

Before You Go

Ask your colleagues for personal contacts in the regions you are headed for, and get in touch with those people when you arrive—you may need their help later. Small gifts are always appreciated (recommended: objects bearing your institution's name and logo, such as coffee cups or hats). Consult a good guidebook or the internet for information about the region you are visiting. It is always useful to have a bit of historical and cultural perspective on the place you will be working.

What Should You Carry in Your Carry On?

Passport, identification that shows your institutional affiliation (e.g., institutional identification card, letter from the museum director on letterhead, business cards), toothbrush, toothpaste, aspirin, prescription medications, change of socks and underwear (in case your baggage does not arrive with you), luggage keys, money, camera, field book, any small expensive objects (such as a GPS device, binoculars, or digital recorder), reading matter, and of course your airline tickets.

What Should You NOT Carry in Your Carry On?

Anything prohibited by the airlines or international regulations, including firearms, explosives, live animals, matches, lighters, fluids (including drinking water), killing agents for specimens, syringes and needles, knives, scissors, or any sharp tools.

CAMP GEAR

bottle opener
candle holders
candles
can opener
chair (folding)
coffee pot (for boiling water)
cook stove, portable (recommended: propane)
cooking utensils
cup (for drinking; recommended: cup with lid)
detergent (dish washing)
dinnerware (plates, glasses, mugs, forks, spoons)
dishcloth
eating utensils (forks, spoons)
entrenching tool (folding shovel)
file to sharpen machete
filter system for drinking and cooking water (Level II system with microbiological micron rating of 0.2 to 1.0)
flagging, plastic (safety orange, for marking trails)
ground cloth for tent
hammer or ax for driving in stakes

hammock
 knife
 lantern (recommended: propane or LED powered by batteries)
 lantern mantles
 lighter, disposable
 machete
 machete scabbard
 matches
 measuring spoons
 mosquito netting (even if sleeping indoors, sleep under mosquito netting,
 especially if indoors is under a thatch roof)
 nylon cord (to secure tents and tarps, hang bags)
 plastic bags, very large
 plastic bags, small (e.g., zipper type bags)
 polyethylene sheeting (to cover gear while traveling, etc.)
 potato peeler
 propane tank for lantern
 propane tank for stove
 rope
 saucepan (two quart) and lid
 scouring sponge
 skillet
 small broom
 spatula
 table, folding
 tape, box sealing
 tape, duct
 tape, electrical
 tape, masking
 tarp, nylon
 tent
 tent stakes (two extra)
 toilet tissue (usage is generally about 0.3 roll per person per day)
 water jug

PERSONAL ITEMS

airline tickets
 alarm clock (battery operated)
 baby powder (to dry feet)
 backpack
 basin plug, universal (for plugging hotel sinks)
 batteries for all battery-operated devices
 battery charger (solar battery charges are available for working in areas
 without electrical supply)
 belt
 belt, utility
 blouse (conservative, for wearing when you apply for permits; select a
 material that dries quickly and does not need ironing)
 books for recreational reading (recommended: inexpensive editions with
 larger type for reading in dim light)
 boot inserts

boots, hiking (break in before the trip)
boots, rubber (with good support; break in before trip)
calculator (small)
canteen
cell phone (many cell phones will not work outside of the country in which they were issued; some service providers require that you make special arrangements before traveling in order to use your cell phone in another country; check with your cell phone provider well in advance of traveling)
clothes line
clothes pins
coat (recommended: water repellent; avoid military-style clothing)
comb
contact lens case
contact lens cleaning supplies
cot
cotton sheets
cotton swabs (Q-Tips)
credit cards
day pack (do not wear a day pack in the city—pickpockets can easily remove things from them)
deck of cards
dental floss
deodorant
dress clothes (conservative, for wearing when you apply for permits, present lectures, meet with important officials)
ear plugs (both to protect your hearing on noisy city streets and to help you sleep)
electrical converter (if traveling in areas without 110 volt current)
emery board (nail file)
eyeglasses
eyeglass repair kit
extra pair of eyeglasses
face mask (for sleeping in rooms that are too bright)
facial tissues
fanny pack (do not wear fanny packs on your butt in the city; wear them in front only)
flashlight (recommended: LED light with ultraviolet option)
flip-flops (shower shoes)
Frisbee (flying disc)
hair brush
hand cream
handkerchief or bandana
hand wipes (moist towelettes, anti-bacterial; for cleaning up before eating or while traveling)
hat for rain
hat for sun
insect repellent containing DEET (diethylmetatoluamide) in liquid form or as pump spray
jacket, light weight (avoid military-style clothing)
keys, extra, to anything you have which locks (have another member of your party carry your extra keys)

knife, hunting
 knife, folding (e.g., Swiss Army knife)
 laces (extra) for boots and shoes
 laundry bag
 mirror, small (recommended: stainless steel)
 money belt or money pouch
 nail brush (useful for both fingernails and for clothing)
 nail clippers
 neck pillow (inflatable)
 pants (of material that dries quickly, e.g., cotton or khaki)
 passport (make sure that it will be valid throughout the duration of your trip)
 photo ID (e.g., valid driver's license, institutional identification card)
 photographs of family and home (great to start conversations with people
 you meet)
 pillow
 pillow case (stuff it with extra clothing to make a pillow)
 poncho, water repellent
 pajamas
 radio that receives AM, FM, and short-wave (recommended: C. Crane radios)
 raincoat
 razor
 razor blades
 safety pins (assorted sizes)
 sanitary napkins
 sewing kit (with extra buttons)
 shampoo
 shoes
 shorts
 shirts, long sleeve (of material that dries quickly; avoid military-style
 clothing)
 shirts, short sleeve (of material that dries quickly; avoid military-style
 clothing)
 skirt (conservative, for wearing when you apply for permits)
 sleeping bag
 sleeping pad
 soap
 soap dish
 shoes, city
 shoes, walking/hiking
 socks, cotton
 socks, woolen
 sunglasses
 sweater
 swimsuit
 toothpicks
 T-shirts
 toothbrush
 toothbrush case
 toothpaste
 towels
 umbrella

underwear
Vaseline
washcloth
watch
windbreaker

TOOLS

Carefully consider the equipment you are taking and bring along any specialized tools, such as Allen wrenches or torx drivers, to do whatever repairs you will be capable of doing in the field.
hose clamps (adjustable, variety of sizes)
multi-tool (purchase a decent quality multi-tool; cheap tools don't hold up with use)
oil
pliers, needle-nosed (with wire cutter blade)
pliers, regular
screwdriver set, slot and Philips (recommended: one handle with multiple blades)
selection of a few small screws, bolts, nuts, and washers to repair suitcases, travel boxes, and other items
vice grips (small)
WD-40 (small container)

FIRST AID AND MEDICINES

adhesive tape (surgical tape)
acetaminophen
aloe vera cream for minor rashes, irritations, burns and scrapes
antacid tablets
antibacterial cream
antibiotics (e.g., ampicillin, tetracycline)
antidiarrheal [recommended: Lomotil tablets (diphenoxylate), Imodium (loperamide), Pepto Bismo (bismuth subsalicylate)]
antihistamine (with chlorpheniramine maleate)
anti-malarial drugs (these can be hard on the liver and may damage hearing if taken long time; so make certain you need them, and consult with your local health center or the Center for Disease Control for which type to take.
aspirin
baking soda (use to make a paste to relieve itching, soothe minor burns, or use 1/2 tsp in 1 oz of water as an antacid)
bandaids (sticking plasters)
bandage tape
Betadine
charcoal (for stomach gas)
Chiggerex
cloth sling
decongestant (with pseudoephedrine)
dental floss
Dramamine (dimenhydrinate, for motion sickness)
ear drops
Elastoplast strip
ethyl alcohol (95%, for sterilizing)

emery board (can also be used for filing chipped or broken teeth)
 eye wash
 flea powder
 foot powder, anti-fungal (e.g., Desenex or Tinaderm)
 gauze compresses, sterile
 gauze pads, sterile
 ginger root capsules, 400 to 500 mg (for stomach gas)
 hepatitis vaccination
 ibuprofen
 lip balm
 menstrual pads (for use as compresses)
 moleskin, adhesive
 oral analgesic containing benzocaine (ethyl p-aminobenzoate)
 skin cream for rashes and insect bites (containing benzocaine and/or aloe vera)
 snake bite kit (e.g., Extractor™ from Sawyer)
 sulfur powder (mix 50:50 with baby powder for chiggers)
 sun block
 suture thread (sterile)
 tension bandage
 thermometer
 vitamin C (to ameliorate colds and flu)
 vitamins, multiple
 tetanus shot (renewed every 5–10 years)
 Tiger Balm (external analgesic with methyl salicylate and menthol)
 topical antiseptic
 zinc oxide ointment (for sunburn or skin rash; e.g., Desitin)

Tips

- In many countries you can buy antibiotics and prescription strength pain killers over-the-counter.
- Carry your medications in a proper medicine bottle (e.g., a prescription medicine bottle) so they do not look like contraband.
- If you get diarrhea, take steps to avoid dehydration. The World Health Organization recommends this formula: to one liter of clean water, add 3.5 g sodium chloride, 1.5 g potassium chloride, 20.0 g glucose, and 2.9 g trisodium citrate. A simpler formula if you cannot get those ingredients is to add 1/2 tsp salt and 4 tbs sugar to one liter of clean water. Follow any diarrheal episodes by eating yogurt (once the diarrhea stops) to restore intestinal flora and fauna.
- Check on needed immunizations before your trip, particularly hepatitis, polio, typhoid, tetanus, and yellow fever.

GENERAL FIELDS AND COLLECTING GEAR

address book with mailing addresses, telephone numbers, FAX numbers, and email addresses of your home institution, critical contacts, and relevant professional colleagues
 altimeter
 binoculars
 camp chair, folding
 camera batteries, extra
 compass

dictionary, of appropriate foreign language
dissecting kit
envelopes, large (for receipts, permits, etc.)
eraser (for pencil)
field book
field book paper (acid free or spun-bonded polyethylene)
field box (recommended: high impact molded plastic, with locks)
file (for sharpening machete)
flash light, large (recommended: LED)
flashlight, small (recommended: LED)
global positioning device (GPS), with extra batteries
institutional envelopes and address labels
institutional letterhead
keys, extra, for all locks
logbook for receipts
maps
marker, permanent (for labeling gear; fine point and blunt point Sharpie
 markers recommended)
measuring tape
mesh bags, nylon (for food and other supplies in camp)
notebook, pocket size (for carrying on the trail and in the city)
padlocks for baggage (including field box)
pen, disposable (with archival quality ink)
pen, technical
pencil, no. 2 lead
pencil, mechanical (with HB lead)
pencil sharpener
photo scale (for placement in photos)
plastic tape in various colors
sharpening stone (for pocket knife)
sketchbook
sleeping bag (select weight and till carefully, based on destination)
stool (folding)
stopwatch
thermometer, maximum/minimum
thermometer, pocket case
thermos for liquid nitrogen
psychrometer
rain gauge
references for field identifications
aluminum foil
aquarium net

TISSUE SAMPLING GEAR

bleach (for cleaning tools before taking samples)
buffer solution to back up frozen samples
catalog (for tissue samples)
cryo marker
cryo tubes
cryo tube holder
cryo tube marking pen

dissecting needle (for scratching identification marks on cryo tubes)
 ethanol, 95%
 ethanol, absolute
 eye dropper (for filling tubes with ethanol)
 ice chest
 forceps (keep clean for use with tissues)
 Kimwipes
 liquid nitrogen tank
 liquid nitrogen (check availability before leaving on trip)
 nylon stockings
 paper towels
 plastic tubes, large (heavy duty, to fill with water and freeze in nitrogen tank
 fix return trip)
 scalpel (keep clean for use with tissues)
 scalpel blades (keep clean for use with tissues)
 scissors (keep clean for use with tissues)
 strainer
 string
 tongs

HERPETOLOGY GEAR

benzocaine (ethyl p-aminobenzoate) based oral analgesic (e.g., Oragel)
 blowgun
 blowgun darts
 bottle forceps
 buffer for formaldehyde solutions (pre-measured capsules of dibasic and
 monobasic salts)
 calipers
 cheese cloth
 Chloretone, 1M solution (hydrous chlorobutanol)
 chloroform
 cloth bags (variety of sizes)
 dip net
 ethyl acetate
 ethyl alcohol
 fishhooks
 fishing pole, collapsible (for noosing lizards)
 forceps, bottle
 forceps, placental
 forceps, small
 formaldehyde-resistant gloves
 formaldehyde
 gallon plastic jar, wide mouth
 gloves, work (recommended: leather palms and fingers, canvas back)
 head lamp (with beam that can be focused; recommended: LED)
 head lamp bulbs (extras)
 Lidocaine
 microphone, for tape recorder
 microphone batteries
 monofilament
 MS-222 (tricaine methanesulfonate)

Nembutal (aqueous sodium pentobarbital; bring a copy of your permit to possess this chemical)
paper towels
plastic bags, large
plastic bags, medium
plastic bags, small
pocket magnifier (6x–10x)
pocket microscope (30x)
preserving trays, plastic, with snap-on tops
recorder, digital
recorder batteries
rubber bands, very large (for collecting specimens)
ruler (small)
scales, electronic (portable)
scales, spring
scalpel blades
scalpel handle
scissors, bandage
scissors, small
sewing needle (for tagging snakes, etc.)
slingshot
slingshot ammunition
snake hook
snake tongs
stump ripper
syringes and needles (do not pack these in your personal gear)
tags, blank
tags, field series (pre-numbered)
tag string
thread
tricaine methanesulfonate
turtle traps
vials (screw cap; large selection of sizes)
work tray

APPENDIX III. SOURCES OF SUPPLIES AND INFORMATION**FIELD GEAR**

- Ben Meadows—variety of equipment including portable electronic scales.
- BioQuip—mostly entomological gear, but also sticky trap adhesive and a good selection of vials, forceps, etc.
- Cabela's—hunting and camping equipment, including headlamps.
- Consolidated Plastics Company—plastic bags, containers, shipping containers, field boxes.
- Forestry Suppliers—professional outdoor and environmental supplies.
- Integra Miltex—surgical instruments such as placenta forceps with serrated ring jaws.
- Nichols Net and Twine, Inc.—netting, seines.
- REI—camping and outdoor equipment.

Ben Meadows
 PO Box 5277
 Janesville, Wisconsin 53547-5277
 800-241-6401
www.benmeadows.com

BioQuip
 2321 Gladwick Street
 Rancho Dominguez, California 90220
 310-667-8800
www.bioquip.com

Cabela's
 800-237-4444
www.cabelas.com

Consolidated Plastics Company, Inc.
 4700 Prosper Drive
 Stow, OH 44224
 800-362-1000
www.consolidatedplastics.com

Forestry Suppliers
 205 West Rankin Street
 P.O. Box 8397
 Jackson, Mississippi 39284-8397
 800-647-5368
www.forestry-suppliers.com

Integra Miltex
 589 Davies Drive
 York, Pennsylvania 17402
 800-645-8000
www.miltex.com

Nichols Net and Twine, Inc.
2200 Highway 111
Granite City, Illinois 62040
800-878-6387
www.nicholsnetandtwine.com

REI
800-426-4840
www.rei.com

HERPETOLOGICAL COLLECTING GEAR

Midwest Tongs, Inc.
14505 Harris Road
Greenwood, Missouri 64034
877-878-6647
www.tongs.com

Pillstrom Tongs
903 S 10th Street
Rogers, Arizona 72756-5211
479-452-3001
www.pillstromtongs.com

Pondside Herp & Pet Supply
2405 Stonebridge Circle Suite 15
West Bend, Wisconsin 53095
262-429-1121
www.herpsupplies.com

Whitco Manufacturing, Inc.
301 W 6th Street
Weatherford, Texas 76086
817-594-2142
www.whitneysnaketongs.com

Plastic Bags, Heat Sealers, HDPE Buckets, Shipping Boxes

Consolidated Plastics Company, Inc.
4700 Prosper Drive
Stow, OH 44224
800-362-1000
www.consolidatedplastics.com

Gaylord
PO Box 4901
Syracuse, NY 13221-4901
800-448-6160
<http://www.gaylord.com/c/Natural-History-Preservation>

Uline
12575 Uline Drive
Pleasant Prairie, Wisconsin 53158
800-295-5510
www.uline.com

Preserving Trays

I recommend good quality polypropylene trays, such as Rubbermaid brand. Quality polypropylene trays will be marked with the letters “PP,” and are available at houseware and hardware stores, as well as from plastic container companies.

Snakebite Kit (caution: see comments in section on Snakebite)

The Extractor
Sawyer Products
PO Box 186
Safety Harbor, Florida 34695
800-356-7811
www.sawyer.com

Recording Equipment Reviews and Recommendations

Cornell Laboratory of Ornithology Macaulay Library
macaulaylibrary.org

Wildlife Sound Recording Society
www.wildlife-sound.org

COLLECTION MANAGEMENT SUPPLIES

Fixatives and Preservatives

Contact chemical suppliers serving your area.

Alcohol Testing Equipment (digital density meters, hygrometers)

Cole-Parmer
625 East Bunker Court
Vernon Hills, Illinois 60061
800-323-4340
www.coleparmer.com

Fisher Scientific
300 Industry Drive
Pittsburgh, Pennsylvania 15275
800-766-7000
www.fishersci.com

Kessler Thermometer Corporation
86 Seabro Avenue
Amityville, New York 11701
631-841-5500
www.kesslerusa.com

Ethyl Alcohol Permits

Alcohol and Tobacco Tax and Trade Bureau
National Revenue Center
1310 G Street NW, Box 12
Washington, DC 20005
202-453-2000
www.ttb.gov

Safety Supplies and Spill Kit Supplies

Fisher Scientific
800-766-7000
www.fishersci.com

Lab Safety Supply
800-472-4643
grainger.com

Test Kits (e.g., EM Science Formaldehyde Test Kit) See Lab Safety Supplies

Conservation Supplies

(acid-free envelopes for photographs and documents, acid-free paper and board, data loggers, Mylar enclosures to archive tags and labels, plastic marking pens, pH testing pens, Pigma pens, Sakura Pigma Graphic pens, Teflon film, Tyvek sheets, UV filters, ethafoam, etc.).

Gaylord
PO Box 4901
Syracuse, NY 13221-4901
800-448-6160

Talas
330 Morgan Avenue
Brooklyn, NY 11211
212-219-0770
<http://www.talasonline.com>

University Products
517 Main Street
Holyoke, MA 01040
800-628-1912
www.universityproducts.com

Containers***HDPE Buckets***

See Plastic Bags, Plastic Tube Bags, Heat Sealers, HDPE Buckets

Gaskets (for glass-top bail jars)

Manufactured Rubber Products
4501 Tacony Street
Philadelphia, Pennsylvania 19124
215-533-3600

Potomic Rubber Company, Inc.
9011 Hampton Overlook
Capitol Heights, Maryland 20743
301-336-7400
www.potomacrubber.com

Jars and Vials

Kols Containers, Inc.
1408 Desoto Road
Baltimore, MD 21230
800-457-5657

Chelsea Bottle Company, Inc.
217 Nahanton Street
Newton, MA 02150
617-969-6061

Carolina Biological Supply
2700 York Road
Burlington, North Carolina 27215
800-334-5551
carolina.com

Dixon Glass Limited
127-129 Avenue Road
Beckenham
Kent BR3 4RX, United Kingdom
00 44 208 778 6458
00 44 7824 422250
<http://www.dixonglass.co.uk/bespoke-glassware/>

Fisher Scientific
www.fishersci.com
800-766-7000

Qorpac (phenol resin and PHEN-PTFE closures)
1195 Washington Pike
Bridgeville, PA 15017
800-922-7558
www.qorpak.com
(also available through major distributors such as Fisher Scientific)

Schott Duran (glass tubes, sealed on one end)
<http://www.duran-group.com/en/products-solutions/laboratory-glassware/service/online-catalogue.html>

Jar Closures (screw-top, flexible polypropylene)

Kols Containers, Inc.
1408 Desoto Road
Baltimore, MD 21230
800-457-5657

Chelsea Bottle Company, Inc.
217 Nahanton Street
Newton, MA 02150
617 -969-6061

Jar Sealing Tape

PPA (polypropylene-acrylic-adhesive) transparent tape
3M Product #5086

Steel Tanks

Delta Designs
PO Box 1733
Topeka, KS 66601
785-234-2244
<http://www.deltadesignsltd.com/>

Steel Fixtures
612 SE 7th Street
Topeka, KS 66601
785-233-8911
<http://www.steelfixture.com/>

Viking Metal Cabinet Company Inc.
24047 W. Lockport Street, Ste. 209
Plainfield, IL 60544
800.776.7767
www.vikingmetal.com

Plastic Bags, Plastic Tube Bags, Heat Sealers, HDPE Buckets

Consolidated Plastics. Inc.
 4700 Prosper Dr.
 Stow, OH 44224
 800-362-1000
<http://www.consolidatedplastics.com/>

Gaylord
 PO Box 4901
 Syracuse, NY 13221-4901
 800-448-6160
<http://www.gaylord.com>

Uline
 12575 Uline Drive
 Pleasant Prairie, Wisconsin 53158
 800-295-5510
www.uline.com

Specimen Tags

Allen Bailey Tag and Label Company
 3177 Lehigh Street
 Caledonia, NY 14423
 585-538-2324
<http://www.abtl.com/>

Storage Supplies***Paper Tubes (for vial storage)***

Jonesville Paper Tube Corporation
 540 Beck Street PO Box 39
 Jonesville, MI 49250
 517-849-9963
www.papertube.com/

Skeleton Boxes

Rigid polystyrene clear plastic boxes

Durphy Packaging Company
 47 Richard Road
 Ivyland, PA 18974
 215-674-1260
www.durphypkg.com/

Althor Products
200 Old County Circle, Suite 116
Windsor Locks, CN 06096
800-688-2693
www.althor.com/

Cardboard boxes, acid free

All Packaging
1515 W 9th Street
Kansas City, Missouri 64191
816-842-3711
www.allpackco.com/

Thermal Printers

Alpha Systems, Inc.
13509 E. Boundary Road
Midlothian, VA 23112
804-744-9870
www.alphasystemsva.com

SOURCES OF COLLECTION MANAGEMENT INFORMATION

American Society of Ichthyologists and Herpetologists (ASIH)

Curation Newsletter, Guidelines for Field Use of Amphibians and Reptiles
<http://www.asih.org/>

Natural Sciences Collection Association (NatSCA)

Journal of Natural Science Collections
<http://www.natsca.org/>

National Park Service

Conserve O Grams and Museum Handbook
<http://www.nps.gov/museum/publications/>

Society for the Preservation of Natural History Collections (SPNHC)

Collection Forum, SPNHC Newsletter
<http://www.spnhc.org>

Standard Symbolic Codes for Institutional Resource Collections in Herpetology and Ichthyology

American Society of Ichthyologists and Herpetologists website
<http://asih.org>; see under Resources tab

Integrated Pest Management (IPM)

Integrated Pest Management Working Group
<http://www.museumpests.net/prevention.asp>

Nhcoll-I—Natural History Collections Listserv

<http://mailman.yale.edu/mailman/listinfo/nhcoll-I>

INSTITUTIONAL ANIMAL CARE AND USE COMMITTEE (IACUC)

Institutional Animal Care and Use Committee Guidebook

grants.nih.gov/grants/olaw/guidebook.pdf

ON-LINE TAXONOMIC SOURCES

SSAR North American Checklist of Scientific and Common Names

<http://ssarherps.org/publications/north-american-checklist/>

Amphibian Species of the World

<http://research.amnh.org/vz/herpetology/amphibia/>

The Reptile Database

www.reptile-database.org

VertNet

<http://portal.vertnet.org>



RECENT PUBLICATIONS OF THE SOCIETY FOR THE STUDY OF AMPHIBIANS AND REPTILES

Breck Bartholomew, Publications Secretary
P.O. Box 58517 Salt Lake City, Utah 84158-0517, USA
Telephone and fax: area code (801)562-2660
E-mail: ssar@herplit.com Web: <http://www.ssarherps.org>

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Shipments outside the USA: Determine the cost for shipments inside USA (above) and then add 6% of the total cost of the order.

CONTRIBUTIONS TO HERPETOLOGY

Book-length monographs, comprising taxonomic revisions, results of symposia, and other major works. Pre-publication discount to Society members.

- Vol. 17. *The Herpetofauna of New Caledonia*, by Aaron M. Bauer and Ross A. Sadler. French translations by Ivan Ineich. 2000. 322 p., 47 maps, 63 figures, 189 color photographs of animals and habitats. Clothbound \$60.00.
- Vol. 18. *The Hylid Frogs of Middle America*, expanded edition, by William E. Duellman. 2001. Review of the 165 hylid species from Mexico through Panama, with paintings by David M. Dennis. Foreword by David B. Wake. 1180 p., 443 figures and maps, 94 plates (46 in color). Clothbound in 2 volumes \$125.00. (Also: separate set of the 46 color plates, in protective wrapper \$45.00.)
- Vol. 19. *The Amphibians of Honduras*, by James R. McCranie and Larry David Wilson. 2002. Comprehensive summary of 116 species, including systematics, natural history, and distribution. Foreword by Jay M. Savage. About 635 p., 126 figures, 33 tables, 154 color photographs of animals and habitats. Clothbound \$60.00.
- Vol. 20. *Islands and the Sea: Essays on the Herpetological Exploration in the West Indies*, by Robert W. Henderson and Robert Powell (eds.). 2003. A collection of essays from 30 herpetologists on their experiences in the West Indies. 312 p., 316 photos, 14 maps. Clothbound \$48.00
- Vol. 21. *Contributions to the history of Herpetology, Volume 2*. by K. Adler, J.S.

RECENT PUBLICATIONS OF THE
SOCIETY FOR THE STUDY OF AMPHIBIANS AND REPTILES

- Applegarth, and R. Altig. 2007. Biographies of 284 leading herpetologists, index to 3512 authors in taxonomic herpetology, and academic lineages of 3810 herpetologists. 465 pp, 270 photographs, Clothbound. \$ 65.00
- Vol. 22. *The Lives of Captive Reptiles*, by Hans-Günter Petzold. 2008. A synthesis of information on captive and wild reptiles (and selected amphibians) covering physiology, behavior, and reproductive biology. 300 p., 63 photographs (57 in color), index. Clothbound \$55.00.
- Vol. 23. *Biology of the Reptilia, vol. 20 (Morphology H)*, by Carl Gans, Abbot S. Gaunt, and Kraig Adler (eds.). 2008. Chapters by three authors cover the skull of the Lepidosauria (lizards, snakes, and tuatara). 769 p., 214 figures, indices. Clothbound \$70.00.
- Vol. 24. *Biology of the Reptilia, vol. 21 (Morphology I)*, by Carl Gans, Abbot S. Gaunt, and Kraig Adler (eds.). 2008. Chapters by five authors cover the skull and appendicular locomotor apparatus of the Lepidosauria (lizards, snakes, tuatara, and amphisbaenians). 791 p., 151 figures, indices. Clothbound \$70.00.
- Vol. 25. *Biology of the Reptilia, vol. 22 (Comprehensive Literature of the Reptilia)*, by Carl Gans and Kraig Adler (eds.), and Ernest A. Liner (comp.). 2010. Bibliography of 22,652 references with cross-referenced subject index. Foreword by Harry W. Greene. 1386 p. Clothbound \$130.00.
- Vol. 26. *The Snakes of Honduras: Systematics, Distribution, and Conservation*, by James R. McCranie. 2011. Comprehensive summary of 136 species, including systematics, natural history, distribution, and conservation. Foreword by Jay M. Savage. 724 p., 65 figures, 23 tables, 180 color photographs of animals and habitats. Clothbound \$95.00.
- Vol. 27. *Herpetofauna of Armenia and Nagorno-Karabakh*, by Marine S. Arakelyan, Felix D. Danielyan, Claudia Corti, Roberto Sindaco, and Alan E. Leviton. 2011. Summary of systematics, natural history, distribution, and conservation of 59 species in 17 families. 186 p., 72 figures, 4 tables, 60 color maps, 151 color photographs of animals and habitats, index. Clothbound \$40.00.
- Vol. 28. *A Contribution to the Herpetology of Northern Pakistan*, by Rafaqat Masroor. 2012. Handbook to the amphibians and reptiles of Margalla Hills National Park and surrounding region. Including color-illustrated keys, 109 color photographs of animals and habitats, color distribution maps for each species, bibliography, checklist of amphibians and reptiles of Pakistan, index. 217 p. Clothbound \$45.00.
- Vol. 29. *Contributions to the History of Herpetology, volume 3*, by Kraig Adler, John S. Applegarth, and Ronald Altig. 2012. Biographies of leading herpetologists (with portraits and signatures), index to 5,290 authors in taxonomic herpetology, and academic lineages of 5,562 herpetologists. Worldwide coverage. color frontispiece. Clothbound \$75.00.
- Vol. 30. *Contributions to the History of Herpetology, volume 1 (revised and expanded)*, by Kraig Adler, 1989 (reprinted in 2014 with corrections and annotations to all 3 volumes in series. Worldwide coverage. Clothbound \$40.00.
- Vol. 31. *Herpetology at Kansas: A Centennial History*, by William E. Duellman, 2015. 340pp., 440 photos. Clothbound \$40.00

RECENT PUBLICATIONS OF THE
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FACSIMILE REPRINTS IN HERPETOLOGY

Exact reprints of classic and important books and papers. Most titles have extensive new introductions by leading authorities. Prepublication discount to Society members.

- BARBOUR, T. and C.T. RAMSDEN. 1919. *The Herpetology of Cuba*. Introduction by Rodolfo Ruidal. 200p., 15 plates. Clothbound. \$55.00.
- BOURRET, R. 1941. *Les Tortues de l'Indochine*. Introduction by Indranel Das. 250 p. 48 uncolored and 6 colored plates. Clothbound. \$65.00.
- DUMERIL, A. M. C., G. BIBRON, and A. H. A. DUMERIL, 1836-1854. *Erpetologie Generale ou Histoire Naturelle Complete des Reptiles*. 9 volumes bound in 5 of text (almost 7000 pages), plus an atlas of 120 plates. clothbound. \$275.00
- FERGUSON, W. 1877. *Reptile Fauna of Ceylon*. First comprehensive summary of the herpetofauna of Sri Lanka. Introduction by Kraig Adler. 48 p. \$8.00
- FRANCIS, E.T.B. 1934. *Anatomy of the Salamander*. Forward by James Hanken and historical introduction by F.J. Cole. 465 p., 25 highly detailed plates, color plate. clothbound \$60.00.
- PERACCA, M.G. 1882-1917. *The Life and Herpetological Contributions of Mario Giacinto Peracca (1861-1923)*. Introduction, annotated bibliography, and synopsis of taxa by F. Andreone and E. Gavetti. 550 pp., clothbound. \$55.00.
- SCHWEIGGER, A.F. 1783 - 1821. *The Life and Herpetological Contributions of August Friedrich Schweigger* Edited by Aaron Bauer, Introduction by Roger Bour. The first scientific review of the world's turtles, covering 78 species (24 of them new plus the new genus Chelydra). 390 p. Clothbound. \$30.00.
- SHAW, G. 1802. *General Zoology, vol. 3: Amphibia*. Herpetological section from the first world summary of amphibians and reptiles in English. Introduction by Hobart M. Smith and Patrick David. 1014 p., 140 plates. Cloth bound \$75.00.
- SMITH, A. 1826-1838. *The Herpetological Contributions of Sir Andrew Smith*. A collection of 10 shorter papers including may descriptions of South African amphibians and reptiles. Introduction by William R. Branch and Aaron M. Bauer. 83 p. Paper cover. \$10.00.

HERPETOLOGICAL CONSERVATION

A series of book-length monographs, including symposia, devoted to all aspects of the conservation of amphibian and reptiles. Prepublication discount to Society members.

- Vol 2. *Ecology, Conservation, and Status of Reptiles in Canada*. by C.N.L. Seburn and C.A. Bishop (eds.). 2007. Chapter by 42 authors dealing with the ecology and conservation. Appendices on the conservation strategy and the current status of reptiles in Canada. 256 pp., Clothbound w/ dust jacket. \$ 40.00
- Vol. 3. *Urban Herpetology*, by Joseph C. Mitchell, Robin E. Jung Brown, and Breck Bartholomew (eds.). 2008. 40 chapters and 13 case studies covering all aspects of the topic, world-wide in scope. 586 p., numerous photographs, figures, and tables; index. Clothbound \$75.00.

RECENT PUBLICATIONS OF THE
SOCIETY FOR THE STUDY OF AMPHIBIANS AND REPTILES

HERPETOLOGICAL CIRCULARS

Miscellaneous publications of general interest to the herpetological community. All numbers are paperbound, as issued. Prepublication discount to Society members.

- No. 27. *Lineages and Histories of Zoo Herpetologists in the United States*, by Winston Card and James B. Murphy. 2000. 49 p., 53 photographs. \$8.00.
- No. 28. *State and Provincial Amphibian and Reptile Publications for the United States and Canada*, by John J. Moriarty and Aaron M. Bauer. 2000. 56 p. \$9.00.
- No. 29. *Scientific and Standard English Names of Amphibians and Reptiles of North America North of Mexico, with Comments Regarding Confidence in Our Understanding*, by the Committee on Standard English and Scientific Names (Brian I. Crother, chair). 2000 [2001]. 86 p. \$11.00.
- No. 30. *Amphibian Monitoring in Latin America: a Protocol Manual/Monitoreo de Anfíbios en América Latina: Manual de Protocolos*, by Karen Lips, Jamie K. Reaser, Bruce E. Young, and Roberto Ibáñez. 2001. 121 p. \$13.00.
- No. 31. *Herpetological Collecting and Collections Management*. (Revised ed.) by John E. Simmons. 2002. 159 p. \$16.00.
- No. 32. *Conservation Guide to the Eastern Diamondback Rattlesnake *Crotalus adamanteus**. by Walter Timmerman and W. H. Martin. 2003. 64 p. \$13.00.
- No. 33. *Chameleons: Johann von Fischer and Other Perspectives*. by James B. Murphy. 2005. 123 p. \$13.00.
- No. 34. *Synopsis of Helminths Endoparasitic in Snakes of the United States and Canada*. by Carl H. and Evelyn M. Ernst. 2006. 86 p. \$9.00.
- No. 35. *A Review of Marking and Individual Recognition Techniques for Amphibians and Reptiles*. by John W. Ferner. 2007. 72 pp. \$11.00.
- No. 36. *Society for the Study of Amphibians and Reptiles: A Fifty Year History 1958 to 2007*. by John J. Moriarty and Breck Bartholomew. 2007. 60 p. \$10.00.
- No. 37. *Scientific and Standard English Names of Amphibians and Reptiles of North America North of Mexico, with Comments Regarding Confidence in Our Understanding, Sixth edition*. Brian I. Crother (ed). 2008. 84 p. \$12.00.
- No. 38. *Nombres Estándar en Español en Inglés y Nombres Científicos de los Anfíbios y Reptiles de México / Standard Spanish, English, and Scientific Names of the Amphibians and Reptiles of Mexico, second edition*. E. A. Liner and G. Casas-Andreu. 2008. 162 p. \$16.00.
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